Veterinary Clinical Parasitology

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PARASITOLOGY

by

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Preface to the First Edition

THE CONTROL of disease can be successful only when preceded by accurate diagnosis. Parasitism by animal forms is universal among domesticated animals. The objective of any parasite is to obtain food, shelter, and a chance to reproduce without imperiling the existence of the essential or the intermediate hosts. This is true parasitism.

Crowding, insanitation, inadequate nutrition, or the breeding of animals with low resistance, encourage parasites to multiply or to attack the host. Thus injury or death will follow. This results in parasitic disease (parasitosis) in a clinically detectable form. Many parasitoses may be diagnosed by the gross routine procedures applicable to disease in general, such as inspection and palpation. Laboratory techniques simply increase the accuracy of diagnosis.

Clinical parasitology is one of the branches of clinical pathology. It serves in diagnosis and prognosis; hence it paves the way toward the prevention and treatment of those diseases in which the predisposing or exciting factors are parasites belonging to the animal kingdom.

The purpose of this publication is to assist in the diagnosis of parasitism and of parasitic disease by means of laboratory techniques, and to show by illustrations the more commonly encountered forms, as well as some of those less often seen.

Because of their diagnostic importance, only three groups of parasites are considered in the present publication. If there is a demand for additional sections, they may be added as soon as illustrative material in sufficient quantity becomes available.

EDWARD A. BENBROOK
MARGARET W. SLOSS

August, 1948

Preface to the Second Edition

Laboratory techniques are increasingly used by veterinarians for the diagnosis of animal diseases. The laboratory may provide the diagnosis when history, symptoms, or gross lesions fail to do so. On the other hand, laboratory procedure should never be used as a substitute for keen clinical inquiry and observation.

To increase the usefulness of this book, numerous additions and revisions have been made.

The photomicrographs have been increased from 247 to 271, including the replacement of four. The 190 illustrations in Section 1 have been regrouped for easier reference.

A description of the fluke egg technique has been added. Eighteen illustrations of helminth ova from man are included so that veterinarians may conduct fecal examinations as a service to physicians.

The section on mites has been revised and four new figures added.

The reference lists have been brought up to date.

It is hoped, as time and material permit, more illustrations and more sections will be added to the existing presentation.

EDWARD A. BENBROOK
MARGARET W. SLOSS

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SECTION 1

Fecal Examination in the Diagnosis of Parasitism

THE PROPER examination of the feces will provide evidence of, or an accurate identification for, most of the parasites which inhabit the alimentary canal. Also, certain parasites of the respiratory tract may be diagnosed by fecal examination, because most of the sputum of animals is swallowed (Figs. 29, 30, 65, 66, 78, 79, 92, 93, 118, 119, 161, 162). Mange or scab mites may be licked or nibbled from the skin, thus accounting for their appearance in the feces (Fig. 130). Fecal examination may also reveal, to a limited extent, the status of digestion, as is shown by the presence of undigested muscle (Figs. 143, 144), of starch, or of fat droplets.

Animals may swallow certain objects that resemble parasite forms. These are known as *pseudoparasites;* they include such things as pollen grains, plant hairs, grain mites, mold spores, and a variety of harmless plant and animal debris (Figs. 67, 132 to 138, 141, 142, 171, 172, 189, 190). *Spurious parasites* are encountered in feces. For example, parasite eggs or cysts from one species of host may be found in the feces of a scavenger or predator host as the result of coprophagy (Figs. 131, 139, 140).

Collection of Fecal Samples

Fresh feces should be used whenever obtainable. Old samples may become dehydrated, making suspension difficult; also worm ova or coccidial oocysts may undergo development, hatching, or disintegration to such a degree as to interfere with diagnosis.

Animal owners may submit fecal samples in all sorts of containers, suitable or not suitable. It is suggested that clients be supplied with clean, wide-mouthed, screw-capped or stoppered jars of at least 60 ml. (2 oz.) capacity. One or two wooden tongue blades are convenient for picking up samples, after which they are discarded. Formed droppings may be transported for a few hours when well wrapped in waterproofed paper.

At least several grams of feces should be collected for an examination. Because of the roughage content, larger samples should be secured from herbivorous than from carnivorous animals.

If defecation does not provide sufficient material, it may be taken directly from the rectum, or, defecation may be induced quickly by inserting a suppository made from bar-soap or a paper match from an ordinary book match folder. Plain water enema-samples may be obtained, but the dilution factor makes them undesirable as a rule. Soapy or oily enemas should not be used. Fecal specimens removed from rectal thermometers are seldom satisfactory in quantity.

If fecal material is to be transported for more than a few hours, it must be preserved. A 10 per cent formalin solution may be added to saturate the sample. Refrigeration will also keep samples in good condition for several days.

Fecal samples to be shipped by postal service, express, or by other means, should be enclosed in leak-proof containers. Proper identification of each sample by means of a label or a tag is necessary.

Gross Examination of Feces

Gross examination should always be made for the detection of living or dead worms or for the detection of the segments of tapeworms. Oily or soapy substances in samples will indicate that the microscopic examination will be difficult or even impossible.

Microscopic Examination of Feces

This may include several techniques such as: (A) The simple smear method; (B) Qualitative concentration methods; and (C) Quantitative concentration methods.

A. The *simple fecal smear method* of microscopic examination is better than no examination at all, but it has many disadvantages. It should be used only when very small samples are available or when lack of equipment or time prevents the use of a more accurate technique. The simple smear is carried out as follows:

1. Place a microslide on a small piece of newspaper.
2. Place a drop of tap water on the center of the slide.

3. With a toothpick, or some similar instrument, detach from the fecal mass a small sample, about the size of a grain of wheat.
4. Mix the sample into the drop of water on the slide until the suspension is cloudy, but not too much so to read the newspaper printing through it. By means of a finely pointed forceps, remove any larger bits of debris that may be present.
5. Gently lower a square 18 mm. or 22 mm. glass or plastic coverglass onto the specimen on the microslide.
6. Examine systematically under low power (x 100) of the microscope, using the high dry power (x 400) for the observation of details (Fig. 14).

B. *Qualitative microscopic concentration methods of fecal examination.* Techniques of this type will be of greatest value in routine clinical diagnosis. They will detect most alimentary-canal parasitisms and, in addition, certain of those from the respiratory tract. They may also serve to diagnose skin mange of the dog, fox, and cat (Fig. 130).

The method to be described is reasonably rapid and its usage is increasing in veterinary diagnosis. It is of value particularly in the field of small animal practice, although it may be very useful in the detection of certain parasitisms of horses, cattle, sheep, goats, swine, and poultry. Animal owners are interested, usually, in seeing parasitic forms under the microscope. Animal surgery is made more safe by postponing operations on parasitized patients until such hosts are de-parasitized. Veterinary hospital contamination, and the transfer of many parasite species from patient to patient, may be avoided through the isolation and treatment of those animals whose feces show evidence of a parasite burden.

A parasitized animal not exhibiting clinical symptoms may enter a veterinary hospital. Should parasitism develop to the clinical stage after that patient returns home, the owner may unjustly conclude that the animal acquired the parasites while in the hospital. Routine examination for parasites of all hospitalized patients would avoid such criticism.

Fecal examination methods can, and should, be conducted in such a manner as to avoid contamination of the laboratory. To prevent the dissemination of odors, keep the samples covered as much as is possible. Various commercial products are available for the masking or for the neutralization of odors.

Concentration of parasitic ova or oocysts from feces may be accomplished in a number of ways. All methods depend upon mixing the fecal sample with a liquid, the specific gravity of which is greater than that of most of such forms, yet less than the specific gravity of most of the fecal debris. Thus the parasite forms rise to the top of the flotation fluid by gravity — a process that may be hastened by centrifugation.

Flotation fluids may be of various composition. Those most commonly recommended include heavy solutions of sodium chloride, sucrose (cane or beet sugar), glycerine, zinc sulfate, zinc acetate, sodium nitrate, sodium acetate, or magnesium sulfate. None of these solutions is ideal for this purpose. Glycerine has too high a viscosity, hence flotation is slow. The saline solutions are low in viscosity but they tend to dehydrate and thus distort parasite forms; also they crystallize rather quickly on the microslide. Solutions of high specific gravity (sp. gr. 1.400) will float too much debris, thus defeating the purpose for which they are intended.

MODIFIED SUGAR FLOTATION TECHNIQUE

Sheather (1923) first proposed heavy sugar solution for fecal flotation technique. Our experience has shown that sugar solution (sp. gr. 1.200 to 1.300) is the most satisfactory flotation fluid available for routine qualitative clinical fecal examinations, employing centrifugation. This solution will fail to float most of the ova of tapeworms, flukes, and thorny-headed worms. This is not a serious objection because tapeworm ova usually leave the host enclosed within the worm's segments which may be seen grossly on or in the feces; and, except in certain localities, flukes and thorny-headed worms are not highly important parasites of domesticated animals. A technique for finding fluke eggs in feces will be found on page 16.

PREPARATION OF SUGAR FLOTATION SOLUTION

1. Materials:
   Granulated sugar .... 454 gm. (1 lb. avoir.)
   Tap water .... 355 ml. (12 fluid oz.)
   Liquefied phenol crystals .... 6.7 ml. (1.8 fluid dr.)
2. Place the tap water in the upper half of a double boiler.
3. Dissolve the sugar in the water by stirring. The water in the lower half of the double boiler should be close to the boiling point (do not dissolve the sugar by means of direct heat).
4. Place phenol (carbolic acid) crystals in a small graduated glass cylinder. Dissolve the crystals by immersing and rotating the graduate in water near to the boiling point.
5. Add the required quantity of liquefied phenol to the sugar solution while stirring the latter. The phenol acts as a preservative and prevents the growth of molds.
6. Store the sugar solution in stoppered 8-oz. (237-ml.) bottles.

APPARATUS FOR A QUALITATIVE MICROSCOPIC CONCENTRATION METHOD OF FECAL EXAMINATION (FIG. 1)

1. *The microscope.* Magnifications of approximately x 100 and x 400 are most suitable for fecal examinations. Therefore, the optical equipment should include an 8X or 10X Huyghenian ocular, 16-mm. and 4-mm. achromatic objectives, and a substage condenser of 1.25 numerical aperture. A mechanical stage and a binocular body tube with matched oculars are not essential, but they will save the examiner's time and help to reduce eyestrain. The addition of an oil immersion objective will equip the microscope for all the important clinical procedures that require microscopy.
2. *Lens paper.* This is essential for keeping optical lenses clean. Squares of about 8 cm. (3 in.) may be stored in a covered dish or can. They should be used once, then discarded.
3. *Xylene.* This is the only safe lens-cleaning solvent except water. Xylene should be dispensed from a dropper-bottle.
4. *Microscope lamp.* Daylight should not be relied upon. There are many suitable types of microscope lamps. A simple type

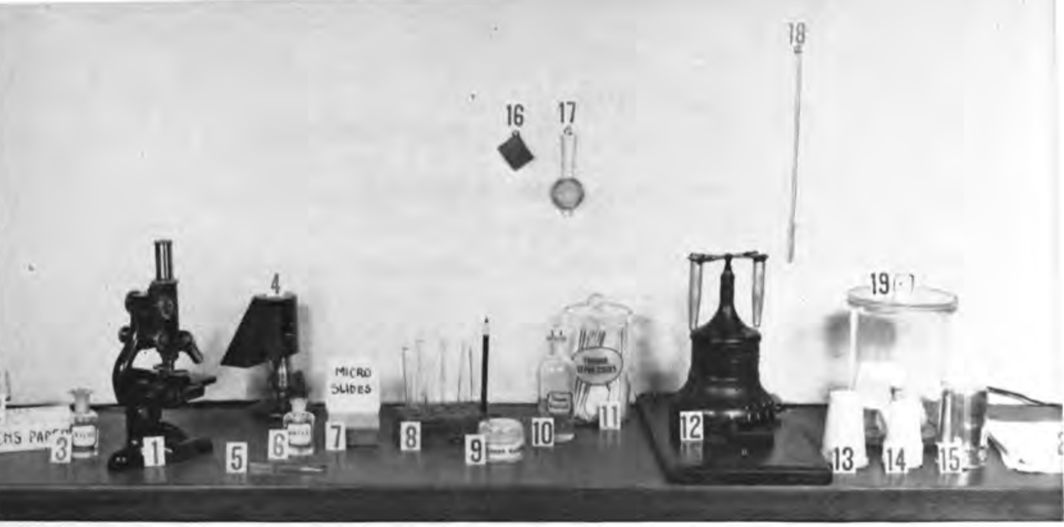

FIG. 1—Apparatus for microscopic examination of feces:

1. Microscope
2. Lens paper
3. Xylene
4. Microscope lamp
5. Coverglass forceps
6. Water-dropping bottle
7. Microslides
8. Test tube block with tubes, headed glass rods, and glass-marking pencil
9. Coverglasses
10. Flotation solution
11. Tongue depressors
12. Centrifuge
13. Large paper cups
14. Small paper cups
15. Aluminum beakers
16. Rubber test tube closure
17. Sieve
18. Test tube brush
19. Jar for waste
20. Towel

to be recommended consists of a metal shade enclosing an inside-frosted 60-watt blue bulb.

5. *Coverglass forceps.* These should always be used when handling micro coverglasses.
6. *Water-dropping bottle.* Any bottle of 30 to 60 ml. (1 to 2 oz.) capacity is suitable, when provided with a medicine dropper. Fresh tap water should be used.
7. *Microslides.* These are the standard 75 x 25 mm. (3 x 1 in.) glass slides. They should be washed and dried before using, and they may be reused repeatedly.
8. *Test tube block with tubes, headed glass rods, and glass-marking pencil.* The test tube block may easily be made by boring 12 mm. (½ in.) holes in a 4 cm. (1½ in.) thick piece of wood. Corresponding 6 mm. (¼ in.) holes are bored to hold

the headed glass rods; and a 10 cm. (3/8 in.) hole is bored to accommodate a glass-marking pencil.

The test tubes recommended are 10 cm. (4 in.) long by 12 mm. (1/2 in.) outside diameter (Fig. 2).

The headed glass rod (Fig. 2) is 5 to 7 mm. (3/16 to 1/4 in.) in diameter by 13 cm. (5 in.) in length. In making the

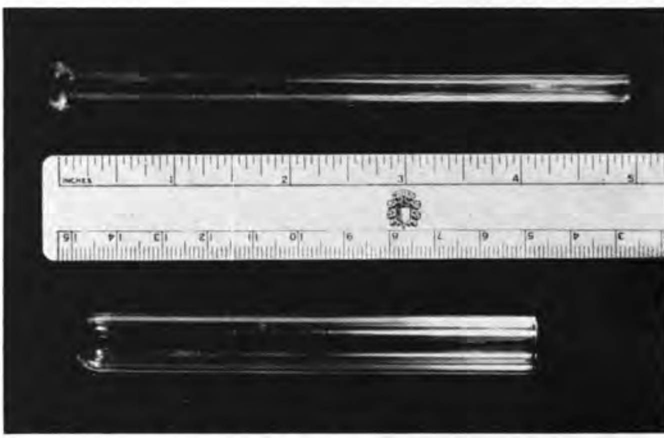

FIG. 2—Test tube **(below)** and headed glass **(above)** shown for comparative size.

head portion, one end of the rod is heated to redness in a Bunsen burner flame. The heated end is then quickly pressed against a warm, flat metal surface such as the head of a hammer, until the head portion of the rod spreads to a diameter of approximately 10 mm. (3/8 in.). After the rod has lost its softness by cooling, it is smoothed by rotating it in the flame. The glass-marking pencil is a standard laboratory item.

9. *Coverglasses.* Any 18 or 22 mm. (3/4 or 7/8 in.) square, glass or plastic coverglass is suitable. The plastic covers are more economical and require no cleaning before they are used, after which they are discarded. Coverglasses should be stored in a covered container such as a small glass dish.

10. *Flotation solution.* The preparation of this fluid has been previously described (page 5).

11. *Tongue depressors.* These are a standard item of wood supplied to the medical professions. They measure 15 cm. long by 2 cm. wide by 2 mm. thick (6 in. by 3/4 in. by 1/16 in.). They are disposable and may be stored for use in a covered glass jar.
12. *Centrifuge.* This instrument may be equipped to hold two or more tubes. The tube holders should accommodate the test tubes (Item 8) as well as the conventional 15 ml. centrifuge tubes. The centrifuge should be provided with a speed regulating switch so that approximately 1,500 revolutions per minute may be maintained. The motor should be of the specifications suitable to the electric current that is available. An electric timer switch, attached to the centrifuge line, may be added in order to shut off the current automatically at the end of the centrifuging period. Angle-type centrifuges are not suitable for the preparation of feces for parasite diagnosis.

13, 14, and 15. *Large paper cups, small paper cups, and aluminum beakers.* Any of these, or similar containers may be used in preparing the fecal samples. The large paper cups have a capacity of 225 ml. (8 oz.). The small paper cups are 90 ml. (3 oz.) capacity; being more economical but less durable than the larger size. It is advisable to discard used paper cups. The aluminum beakers hold 300 ml. (10 oz.). They are comparatively economical because they should last for several years and are easily cleaned.

16. *Rubber test tube closure.* This item may be made from a discarded automobile inner tube, pieces from which are cut approximately 5 cm. (2 in.) square. A hole is punched in one corner so that it may be hung up to dry after it is rinsed clean.
17. *Sieve.* The most suitable sieve is a tea-strainer made of metal wire, approximately 30 mesh to the inch (25 mm.).
18. *Test tube brush.* The brush should be approximately 8 cm. (3 in.) long by 12 mm. (½ in.) in diameter. The bristles should be stiff.
19. *Jar for waste.* Any convenient receptacle having a lid closure may be used. It may contain a disinfectant solution.
20. *Towel.* Smooth cotton or linen towels are used to dry the utensils. Paper towels are a convenience for drying the hands.

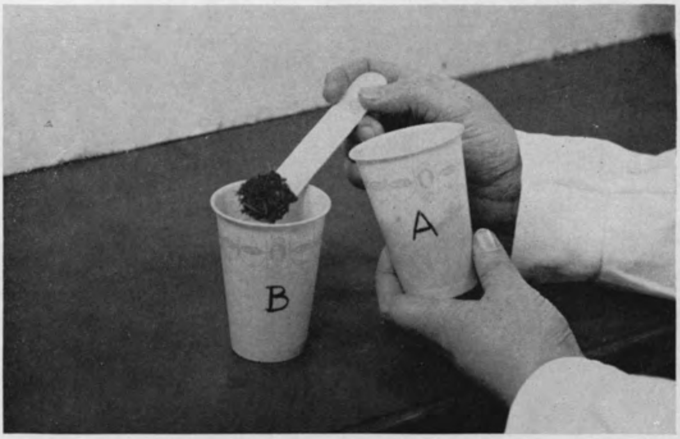

FIG. 3—Transferring approximately 1 gm. of feces from the collecting container **A** to the mixing container **B.**

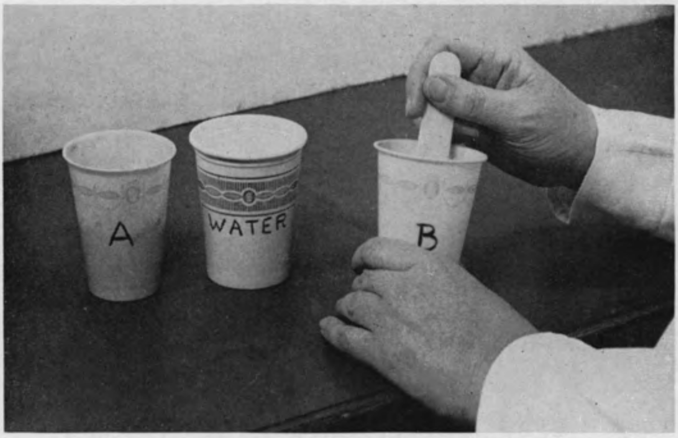

FIG. 4—Suspending the fecal sample in cold water in container **B.**

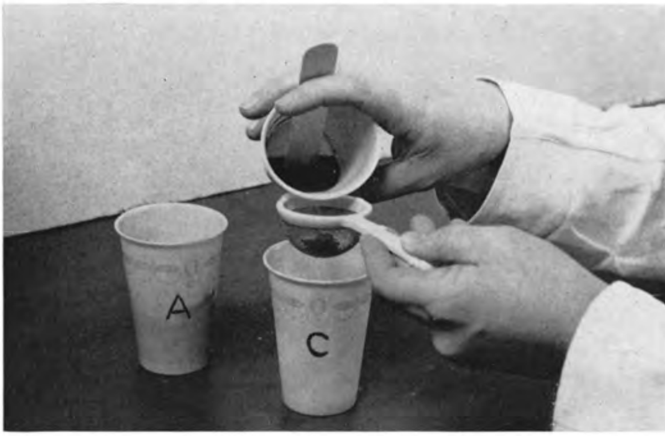

FIG. 5—The watery portion of container **B** is passed through the sieve into container **C.**

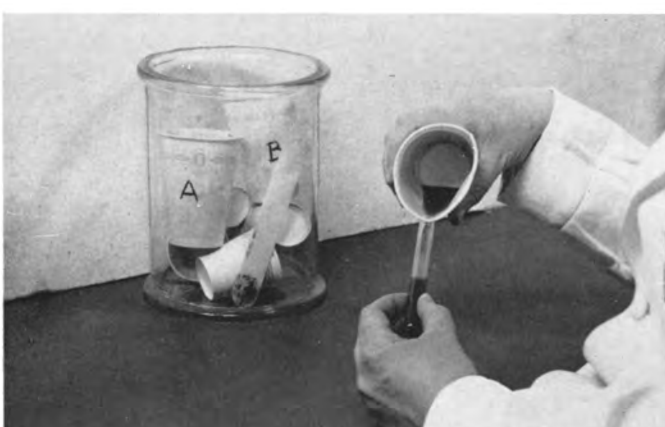

FIG. 6—Transferring sieved feces from container **C** to the test tube. The tube should be slightly less than half filled.

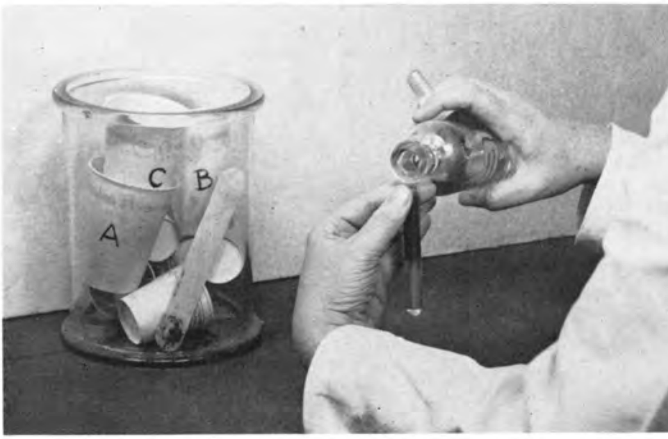

FIG. 7—Adding the flotation solution to the fecal sample in the test tube, leaving about one-fourth inch space at the top of the tube.

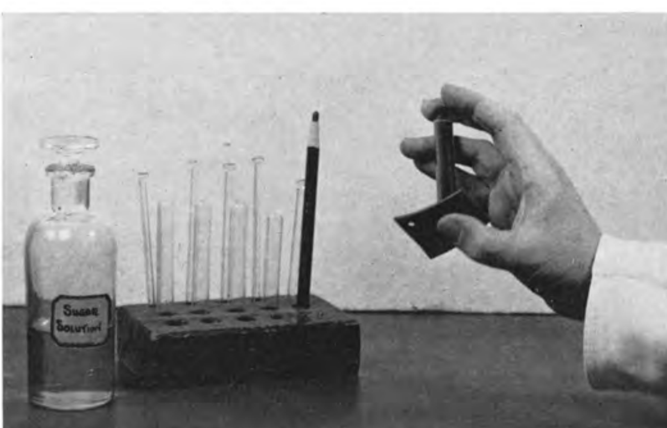

FIG. 8—Mixing the contents of the test tube with the rubber closure applied.

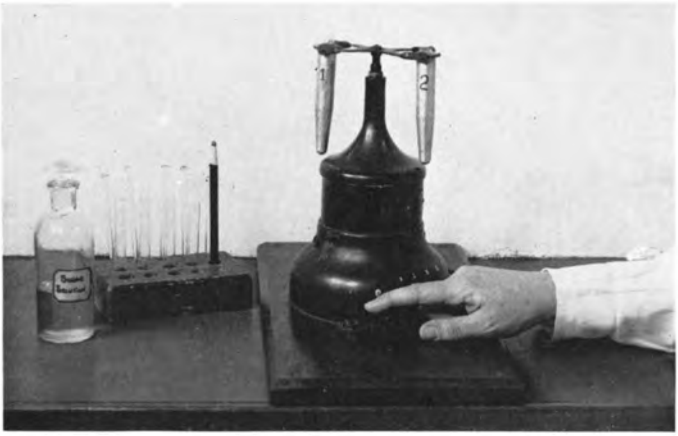

FIG. 9—Centrifuge the sample at approximately 1,500 revolutions for 3 minutes.

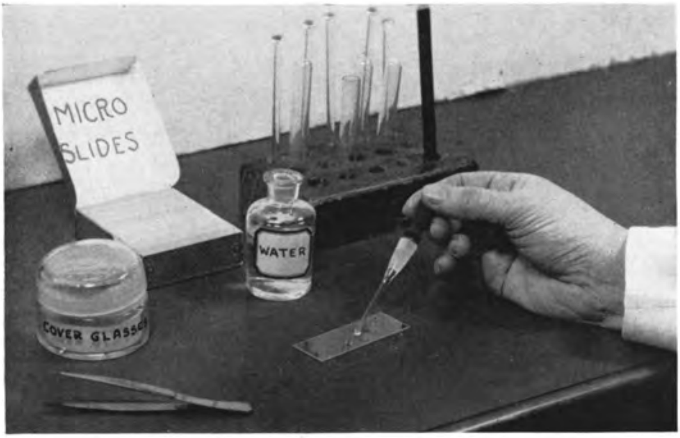

FIG. 10—Placing a drop of water on a microslide.

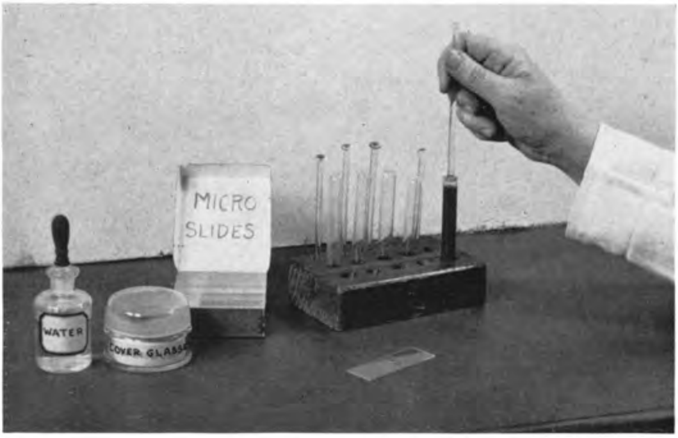

FIG. 11—Removing a drop of fluid, by means of a headed glass rod, from the surface of the centrifuged specimen.

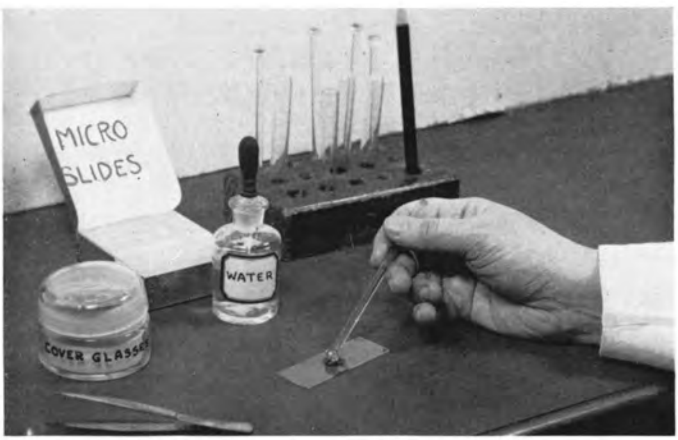

FIG. 12—Transferring the material from the headed glass rod to the drop of water on the microslide.

FIG. 13—Applying the coverglass, using forceps. DO NOT PRESS DOWN!

TECHNIQUE FOR A QUALITATIVE CONCENTRATION METHOD OF FECAL EXAMINATION

1. Transfer approximately 1 gram of feces from the collection container to a mixing cup (Fig. 3).

2. Add small quantities of cold water until stirring results in a watery suspension thin enough to pour (Fig. 4). Too much water will decrease the chance of finding parasite forms.

3. The watery suspension of feces is poured through the sieve into a second container (Fig. 5). The debris left in the sieve is discarded and the sieve is immediately cleaned in running water (preferably hot) before the contents have a chance to dry.

4. The sieved sample is briefly agitated to mix it thoroughly before pouring it into a test tube. The tube should be filled to slightly below the halfway mark (Fig. 6).

5. To the sample in the tube there is added sugar solution to fill the tube to within one-fourth inch (6 mm.) of the top (Fig. 7). Avoid contaminating the opening of the sugar solution bottle.

6. Mix the contents by closing the tube with the rubber thumb protector, then invert the tube some five or six times (Fig. 8). The rubber closure is immediately rinsed off and hung up to dry.

7. Place the tube in the centrifuge. If necessary, place balancing tubes containing water in the centrifuge carrier. Centrifuge the specimen or specimens for three minutes at approximately 1,500 revolutions per minute (Fig. 9). An automatic electric timer switch is very convenient in carrying out this step in the technique.

8. While the centrifuge is in operation, a microslide is placed on the table and a drop of water is centered on it (Fig. 10). Also a clean, headed glass rod, coverglass forceps, and a coverglass are made available. The test tube is transferred from the centrifuge to the test tube holder, care being taken not to agitate the contents.

9. Transfer a drop of sample from the test tube to the drop of water on the microslide (Fig. 11). To do this properly, hold the headed glass rod vertically over the tube, resting the elbow on the table. Slowly lower the head of the glass rod onto the surface of the sample; then quickly withdraw the rod without making contact with the inside of the tube. This operation may require some practice. Then hold the glass rod at about a 45 degree angle and rotate the headed end in the drop of water on the microslide (Fig. 12), thus washing off any parasite eggs or oocysts adhering to the rod. Replace the rod in the test tube block. It should be rinsed and dried before further use.

10. Pick up a coverglass by means of the coverglass forceps. Lower one edge of the coverglass onto the slide near the drop of suspension; then release the forceps as the coverglass is gently lowered onto the drop. The fluid should spread out evenly under the coverglass (Fig. 13). Too rapid an application of the coverglass will probably result in the formation of air bubbles, which may interfere with the microscopic examination of the specimen. *Avoid pressure on the coverglass.*

11. Place the slide on the stage of the microscope so that the near right-hand corner of the coverglass is centered under the low power (16 mm.) objective. Focus on this corner. Adjust the substage condenser and diaphragm of the microscope so as to see a distinct image of the suspension under the coverglass. Using the low power magnification (x 100), systematically move the microslide back and forth until the entire area of the coverglass has been scanned (Fig. 14). Objects having a resemblance to parasite forms may be

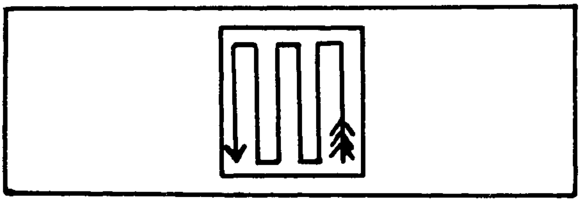

FIG. 14—The material under the coverglass should be systematically examined at 100 magnification according to this diagram.

centered and examined under the high power (x 400) dry lens (4 mm.). Always return to the low power (x 100) lens for further search of the specimen.

If worm eggs and coccidial oocysts are present in the same specimen, the coccidia, being the smaller, tend to float upward until they rest directly beneath the coverglass. Therefore, when the worm eggs are in focus under high power (x 400), the coccidia may be out of focus and vice versa. Both types of parasitic forms may be brought clearly into focus by turning the fine-adjustment knob of the microscope.

MODIFIED FLUKE EGG TECHNIQUE

From the early reference of Cobb (1904) to the latest work of Dennis, Stone, and Swanson (1954), workers have attempted to find a simple, rapid method for demonstrating fluke ova in feces.

Nearly all the investigators have tried some type of flotation technique but were unable to obtain consistent results because of the collapsibility of the ova in solutions of high specific gravity. In the limited number of times we have demonstrated canine lung fluke (*Paragonimus westermanni*) ova in fecal samples, we have used the modification of Sheather's sugar solution technique and have experienced little or no difficulty with the ova collapsing.

The technique of Dennis, Stone, and Swanson (1954) appears to be a relatively simple quantitative method for demonstrating fluke ova. It requires about one-half hour to perform. The following modification of this *quantitative* method is useful for *qualitative* clinical diagnosis.

Reagents for Fluke Egg Technique

1. Detergent solution:
   Liquid detergent ("Joy," or "Glim," or similar) .. 5 cc.
   Tap water ....995 cc.
   1% alum (aluminum potassium sulfate U.S.P.) ... 8 drops
2. Tincture of iodine U.S.P.

Apparatus for Fluke Egg Technique

1. Fecal containers. Samples up to 500 gm. (1 lb.) may be used.
2. Wooden tongue blades for stirring the sample.
3. A tin-coated or zinc-coated funnel, 9 cm. (3½ in.) in diameter with 80 mesh copper screen soldered 25 mm. (1 in.) from the top.
4. Test tubes of 30 cc. (1 oz.) capacity, dimensions 150 x 18 mm. (6 x ¾ in.).
5. Test tube rack or block for holding tubes.
6. Stirring rod (glass or metal), 20 cm. (8 in.) long.
7. Centrifuge tubes, capacity 50 cc. (1.7 oz.).
8. Centrifuge tube rack or block.
9. Wash bottle.
10. Pipette, 2 cc. capacity.
11. Microslides, 75 x 25 mm. (3 x 1 in.).
12. Coverglasses, 22 mm. (¾ in.) diameter.
13. Filter pump (using faucet water pressure, such as the Richards filter pump); or a decanting bottle (using mouth suction); or a bulb syringe of about 30 cc. (1 oz.) capacity.
14. Clinical microscope.

Procedure for Fluke Egg Technique

1. Using a tongue blade, mix the fecal sample thoroughly; and, if it is very dry, add cold tap water to form a pasty mass.

2. Place about 1 gm. of the mixed feces in a 30-cc. (1-oz.) test tube.
3. Add 15 cc. (½ oz.) detergent solution. Mix well with a stirring rod. To avoid sudsing, do not shake.
4. Strain the mixture through the funnel-strainer into a 50-cc. (1.7-oz.) centrifuge tube.
5. Rinse the test tube with more detergent solution and strain.
6. Pour enough detergent solution in a flooding, swirling motion through the feces in the funnel-strainer to fill the centrifuge tube.
7. Allow the tubed mixture to stand for 5 to 15 minutes.
8. Decant three-fourths of the liquid portion from the centrifuge tube.
9. Rewash the fecal material in the funnel-strainer to refill again the centrifuge tube, in order to obtain any ova trapped previously. Discard the funnel contents.
10. Again allow the tubed mixture to stand for 5 to 15 minutes.
11. Again decant all liquid down to about 2 to 3 cc. Do not disturb the sediment.
12. Add 1 to 3 drops tincture of iodine to the sediment, allowing the tube to stand for 2 to 5 minutes.
13. Using a pipette, transfer the sediment to one or more microslides and apply coverglasses.
    (Note: Dennis, Stone, and Swanson recommend placing all of the sediment in a standard Petri dish, adding tap water to make 15 to 20 cc. and searching for ova with a binocular dissecting microscope magnifying 18 x or higher.)
14. Search the sediment on the slide or slides, using a clinical microscope magnifying 100 x.

C. *Quantitative methods of fecal examination.* Various techniques have been proposed for the determination of the *number* of parasite eggs or coccidial oocysts per gram of feces. Such methods are of value in the study of parasite life cycles, or in determining the effects of experimental therapy for the removal of gastro-intestinal parasites. Quantitative fecal techniques are of little value in clinical diagnosis; therefore, such methods are not included in this publication.

References for Section One will be found on pages 169 to 190.

HORSE

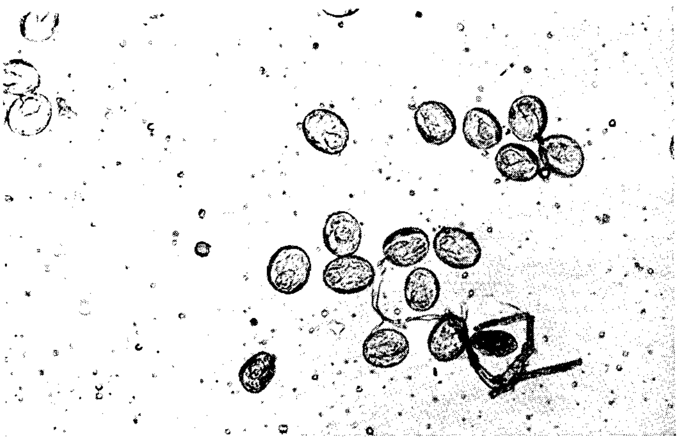

FIG. 15—Ova of **Paranoplocephala mamillana,** the small tapeworm of the horse. x 100.

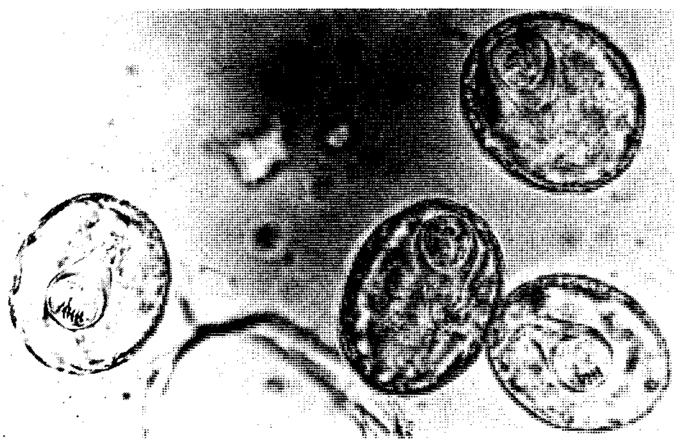

FIG. 16—Ova of **Paranoplocephala mamillana.** The eggs enclose a pear-shaped embryo having six hooklets. x 410.

HORSE

FIG. 17—Ova of **Parascaris equorum,** the ascarid of the horse. The egg shells are rough and thick, and are yellow to brown in color. Also included are three strongyle ova. x 100.

FIG. 18—Ova of **Parascaris equorum.** x 410.

HORSE

FIG. 19—Ova from several species of strongyles of equines. Thirty-nine species of these nematodes have been reported from the large intestine of horses, asses, and mules in North America. The eggs of all species are similar. x 100.

FIG. 20—Ova from two species of strongyles of equines. x 410.

HORSE

FIG. 21—Ova of **Draschia megastoma,** one of the three larger gastric nematodes of the horse. These eggs are elongated; embryonated when laid and are surrounded by a very thin membranous shell. x 100.

FIG. 22—Ova of **Draschia megastoma.** x 410.

HORSE

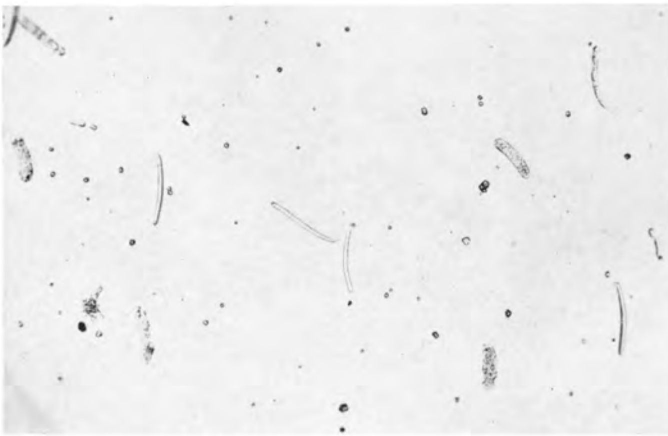

FIG. 23—Ova of **Habronema muscae,** one of the three larger gastric nematodes of the horse. These ova are elongated; embryonated when laid and surrounded by a very thin membranous shell. x 100.

FIG. 24—Ova of **Habronema muscae.** x 410.

HORSE

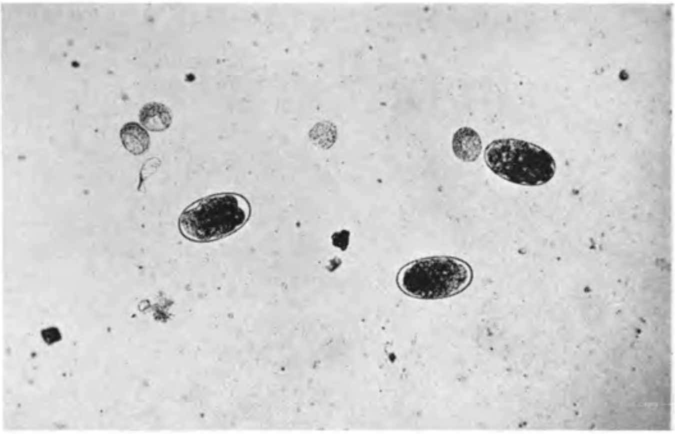

FIG. 25—Ova of **Strongyloides westeri,** the intestinal threadworm of the horse. The three larger eggs are those of strongyles. x 100.

FIG. 26—Ova of **Strongyloides westeri.** These eggs are embryonated when laid. x 410.

HORSE

FIG. 27—Ova of **Oxyuris equi,** the rectal worm of the horse. These eggs may be found in the feces but the examination of anal scrapings is a more accurate method of diagnosis. x 100.

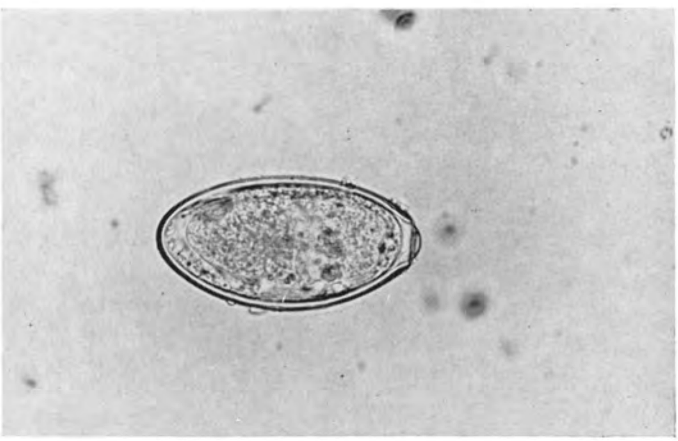

FIG. 28—Ovum of **Oxyuris equi.** Note the operculum (cap) at one end. x 410.

HORSE

FIG. 29—Ova and larvae of **Dictyocaulus arnfieldi,** the lungworm of horses. These were taken from bronchial exudate but they may also be found in feces. The eggs are embryonated when laid. x 100.

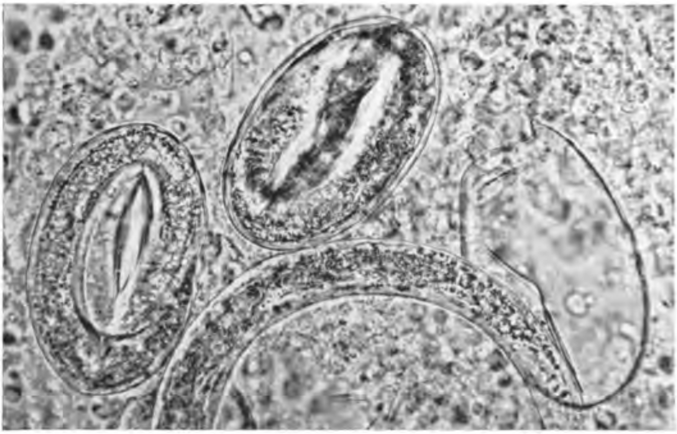

FIG. 30—Ova, part of a larva and an empty egg shell of **Dictyocaulus arnfieldi.** x 410.

CATTLE

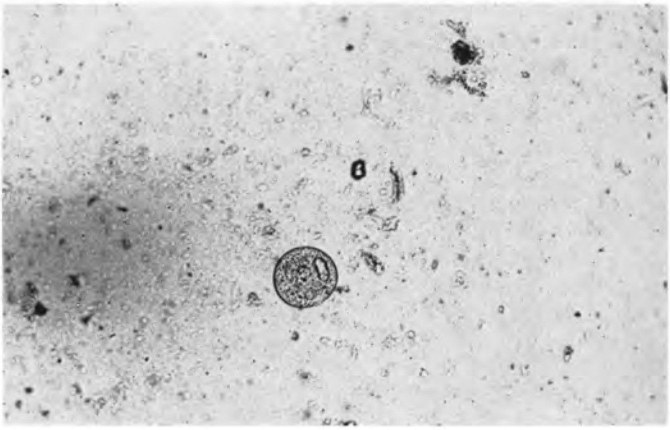

FIG. 31—A cyst of **Buxtonella sulcata** of cattle. This is the resting stage of a large ciliated protozoon of the caecum of cattle. Nothing is known regarding its possible pathogenicity. It is commonly found in cattle feces. x 100.

FIG. 32—**Buxtonella sulcata** cyst. x 410.

CATTLE

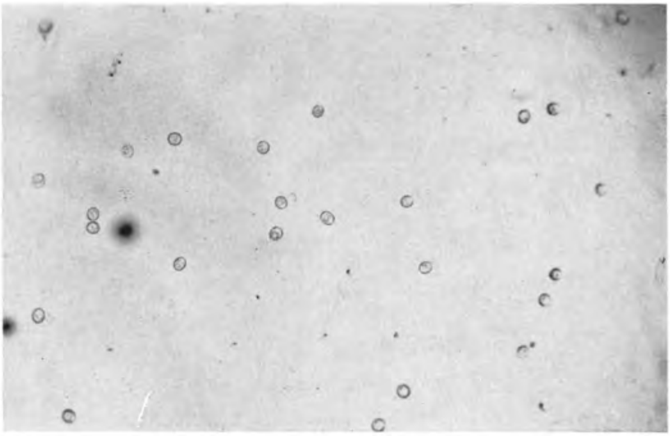

FIG. 33—Oocysts of **Eimeria zurnii**, one of the more pathogenic of the eleven species of coccidia of cattle in North America. x 100.

FIG. 34—Oocysts of **Eimeria zurnii**. x 410.

CATTLE

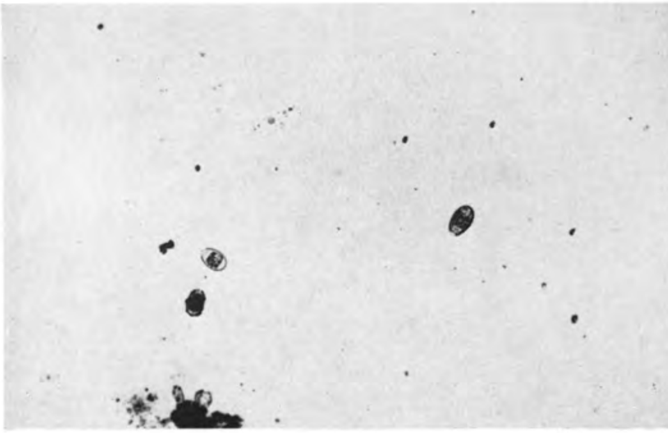

FIG. 35—Oocysts of **Eimeria auburnensis,** a coccidium of cattle. The color is yellowish-brown. One smooth-walled and two rough-walled cysts are shown. x 100.

FIG. 36—Oocysts of **Eimeria auburnensis.** Smooth-walled form at the left; rough-walled form at the right. x 410.

SHEEP, GOAT

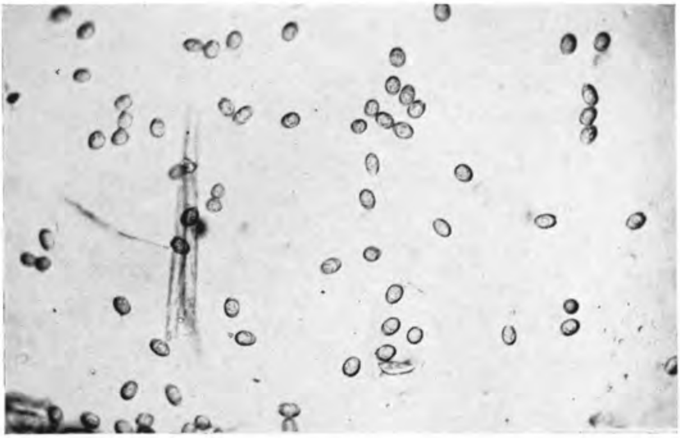

FIG. 37—Oocysts of **Eimeria arloingi,** one of the more pathogenic of the eight species of coccidia of sheep and goats in North America. The color varies from pale yellow to yellowish-green. x 100.

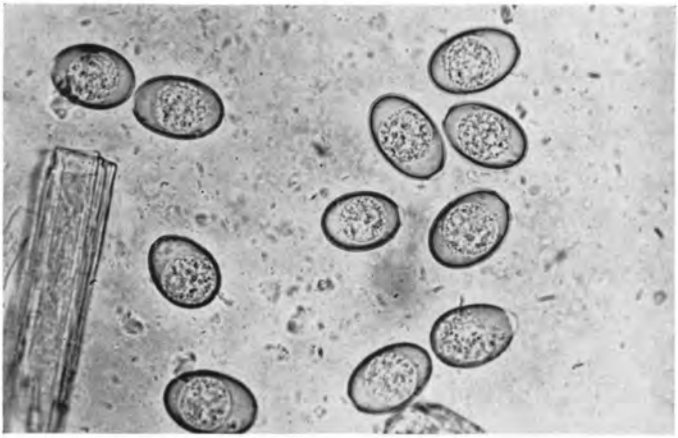

FIG. 38—Oocysts of **Eimeria arloingi.** A polar cap is present at one end of the cyst. x 410.

SHEEP, GOAT

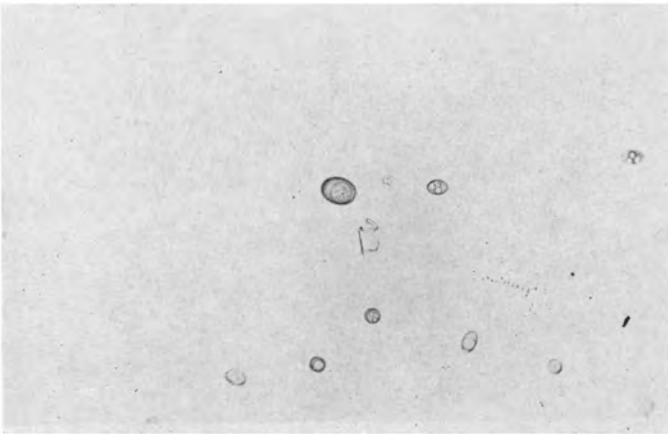

FIG. 39—Oocysts of **Eimeria intricata** and **Eimeria arloingi,** coccidia of sheep and goats. The large oocyst is that of **E. intricata,** the color of which is dark brown. x 100.

FIG. 40—Oocysts of **Eimeria intricata (right)** and of **Eimeria arloingi (left).** x 410.

CATTLE

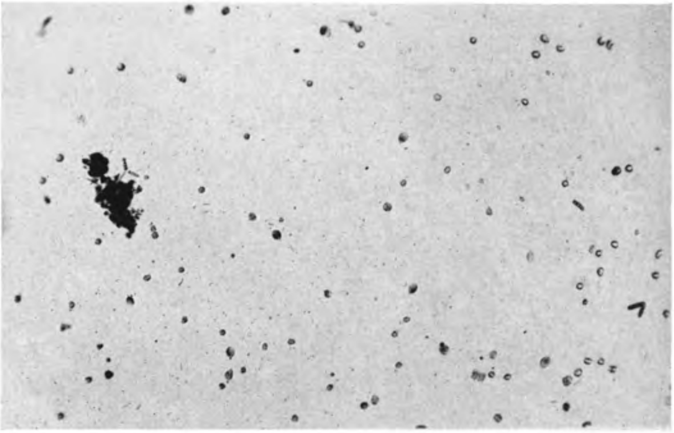

FIG. 41—**Giardia bovis,** a flagellate protozoan of cattle. It is motile. Similar species are found in sheep, goats, dogs, and cats. x 100.

FIG. 42—**Giardia bovis** showing the two posterior flagella. x 410.

CATTLE

FIG. 43—**Giardia bovis** showing the ventral sucking disc and the two nuclei. x 410.

FIG. 44—**Giardia bovis,** oblique view to show the ventral concavity and the posterior flagella. x 410.

CATTLE, SHEEP, GOAT

FIG. 45—Ova of **Fasciola hepatica,** the common liver fluke of cattle, sheep, and goats. x 100.

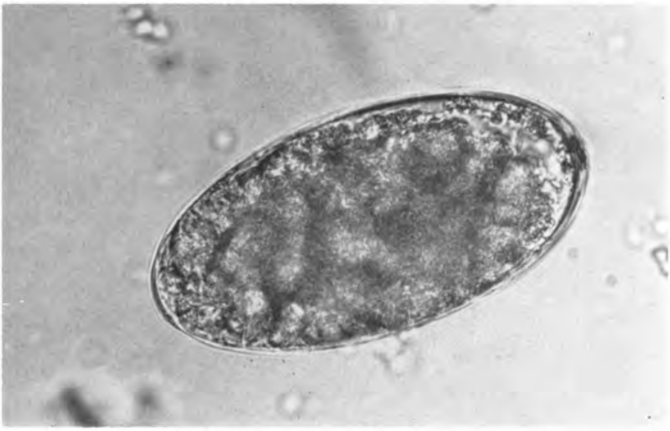

FIG. 46—Ovum of **Fasciola hepatica.** x 410.

CATTLE

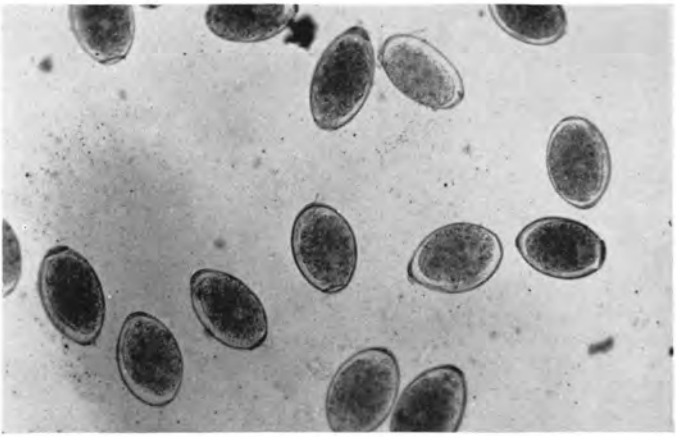

FIG. 47—Ova of **Fascioloides magna,** the large American liver fluke of cattle. The eggs are heavy and sink in sugar solution. x 100.

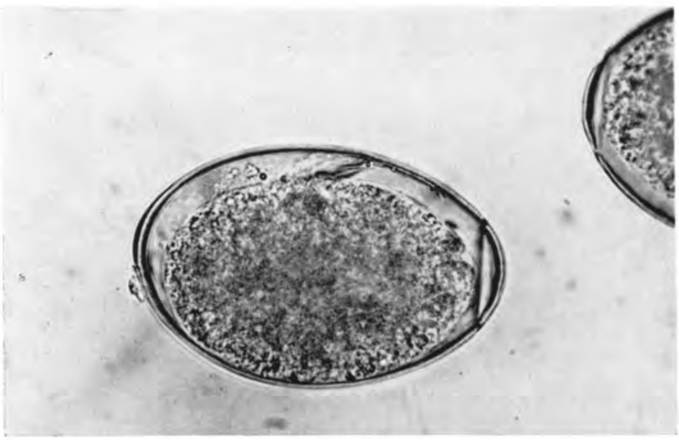

FIG. 48—Ovum of **Fascioloides magna.** Note the operculum at one end. x 410.

CATTLE, SHEEP

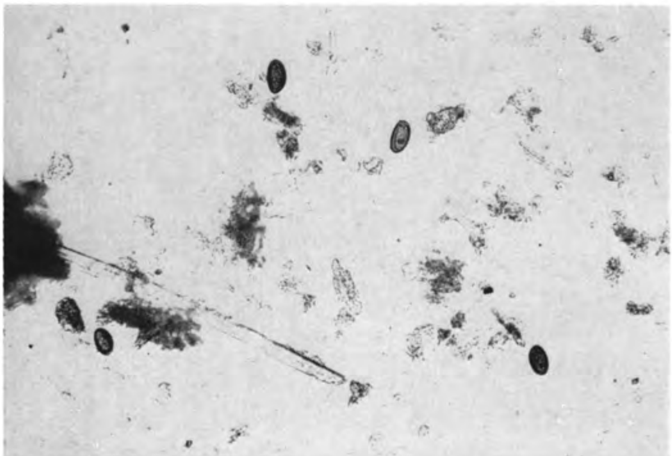

FIG. 49—Ova of **Dicrocoelium dendriticum,** the lancet liver fluke of cattle, sheep, deer, and woodchuck. x 100.

FIG. 50—Ovum of **Dicrocoelium dendriticum.** x 410.

CATTLE, SHEEP, GOAT

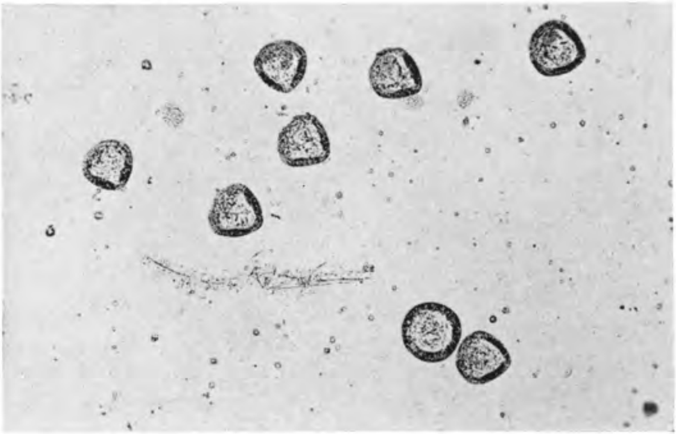

FIG. 51—Ova of **Moniezia expansa,** a tapeworm of cattle, sheep, and goats. x 100.

FIG. 52—Ovum of **Moniezia expansa.** Note the pear-shaped embryo which contains six hooklets. x 410.

SHEEP, GOAT

FIG. 53—A packet containing ova of **Thysanosoma actinioides,** the fringed tapeworm of sheep and goats. These usually leave the host within the tapeworm segments, hence are seldom found on routine fecal examination. x 100.

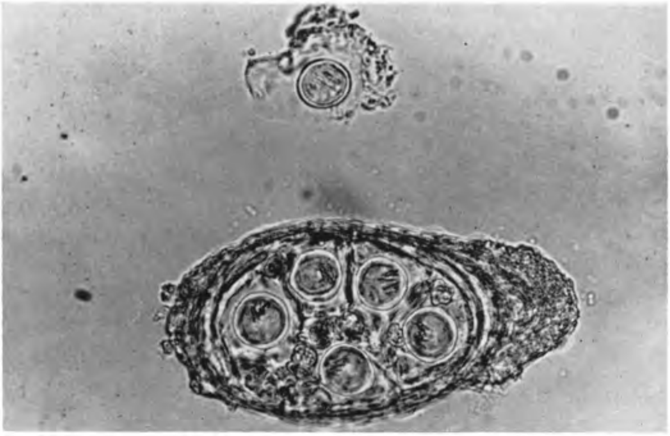

FIG. 54—A packet containing ova of **Thysanosoma actinioides.** Five ova are visible within the packet and one ovum is free. x 410.

CATTLE, SHEEP, GOAT

FIG. 55—Ovum of **Haemonchus contortus,** the common or "twisted" stomach worm of cattle, sheep, and goats. x 100. (See footnote)

FIG. 56—Ovum of **Haemonchus contortus.** x 400.

Note: Cattle, sheep, and goats of North America are reported to harbor 39 species of nematode worms in the alimentary canal. The eggs of the following 24 species are very similar to those seen in Figs. 55 and 56: Common stomach worms (2 species); trichostrongylid worms (4 species); cooperid worms (5 species); nodule worms (3 species); hookworms (2 species); ostertagid stomach worms (7 species); large-mouthed bowel worm (1 species).

CATTLE, SHEEP, GOAT

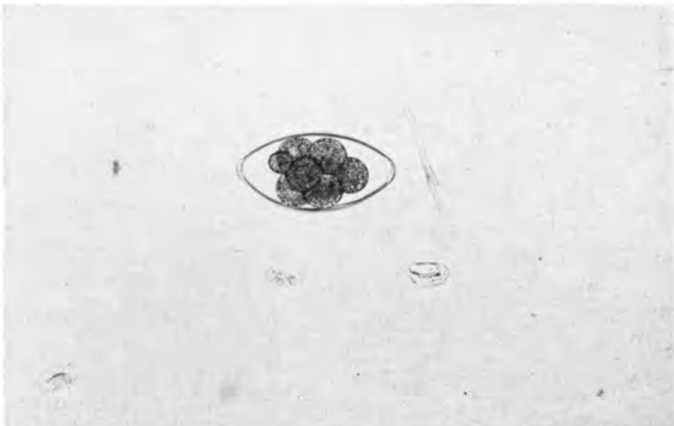

FIG. 57—Ovum of **Nematodirus spathiger,** an intestinal nematode of cattle, sheep, and goats. The two small, embryonated ova are those of **Strongyloides papillosus.** x 100.

FIG. 58—Ovum of **Nematodirus spathiger.** The embryonic mass is in the eight-celled stage. Note the thickened shell at the poles. x 400.

SHEEP, GOAT

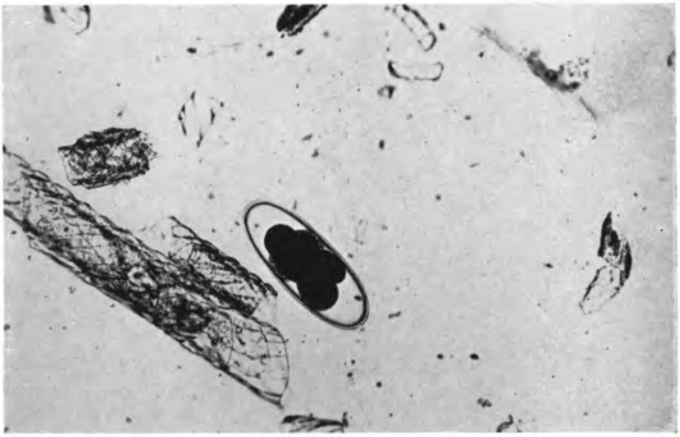

FIG. 59—Ovum of **Marshallagia marshalli,** a stomach worm of sheep and goats. x 100.

FIG. 60—Ovum of **Marshallagia marshalli.** x 410.

CATTLE, SHEEP, GOAT

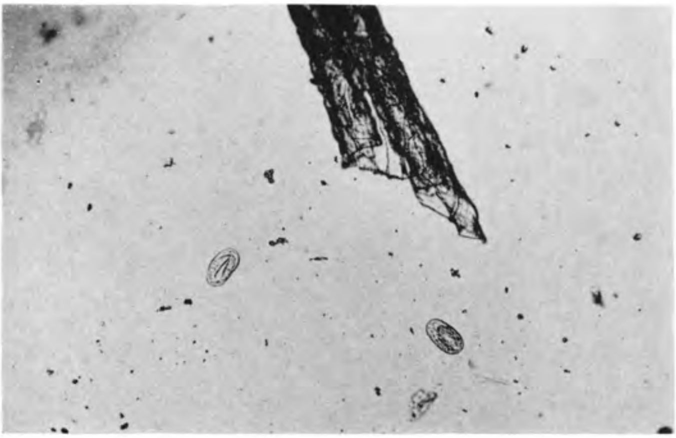

FIG. 61—Ova of **Strongyloides papillosus,** a threadworm of the small intestine of cattle, sheep, and goats. x 100.

FIG. 62—Ovum of **Strongyloides papillosus.** The eggs of this nematode are embryonated when laid. x 410.

CATTLE

FIG. 63—Ova of **Neoascaris vitulorum,** the ascarid of cattle. x 100.

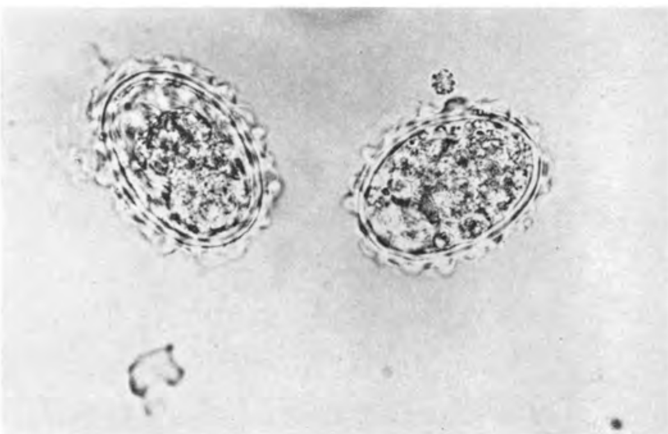

FIG. 64—Ova of **Neoascaris vitulorum.** x 410.

SHEEP, GOAT

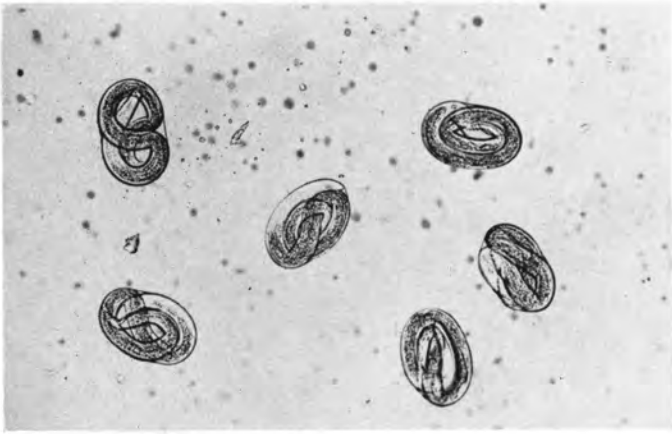

FIG. 65—Ova of **Dictyocaulus filaria,** a lungworm of sheep and goats. These were taken from bronchial exudate, but they may also be found in feces. The eggs are embryonated when laid. x 100.

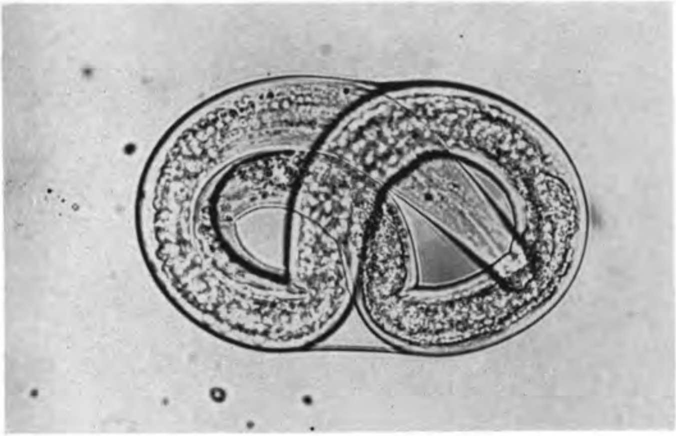

FIG. 66—Ovum of **Dictyocaulus filaria.** x 410.

CATTLE

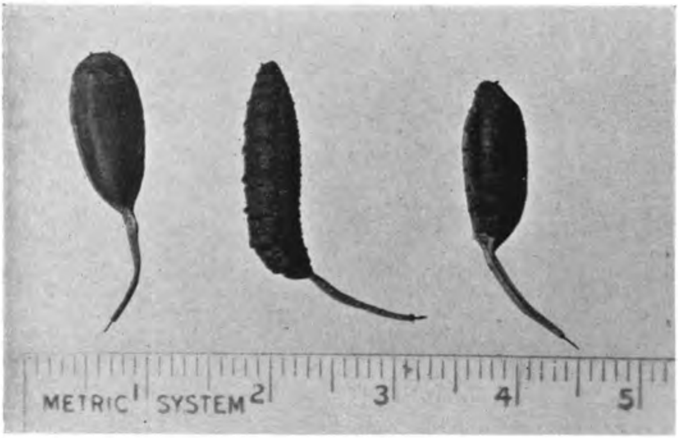

FIG. 67—Pseudoparasite. Rat-tailed maggots from cattle feces. These are the larvae of harmless flies commonly known as drone flies. They belong in the dipterous family Syrphidae. x 1.7.

SWINE

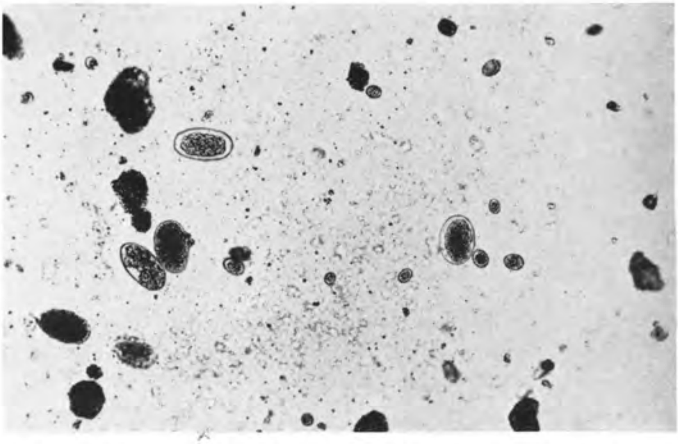

FIG. 68—Oocysts of **Eimeria** sp., coccida of swine. Several species are shown. The ova are those of **Oesophagostomum** sp., one of the nodule worms. x 100.

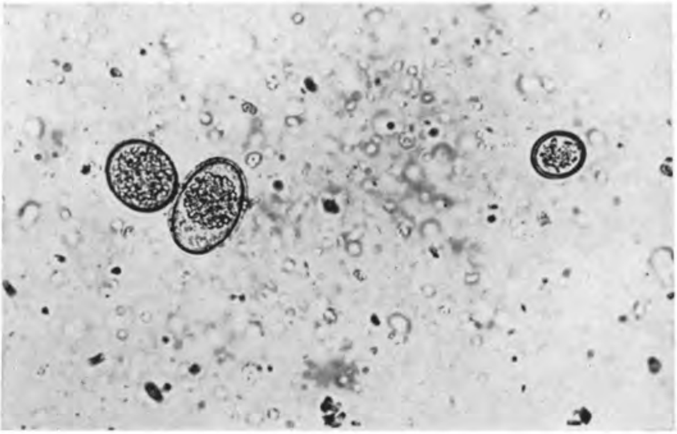

FIG. 69—Oocysts of **Eimeria** sp. Three species are shown. x 410.

SWINE

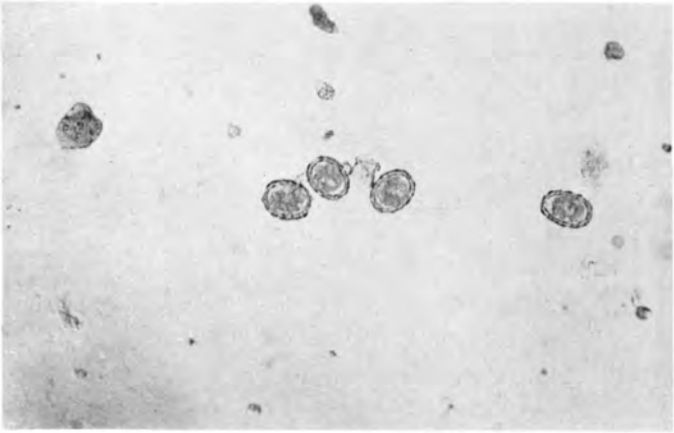

FIG. 70—Ova of **Ascaris lumbricoides,** the ascarid of swine. x 100.

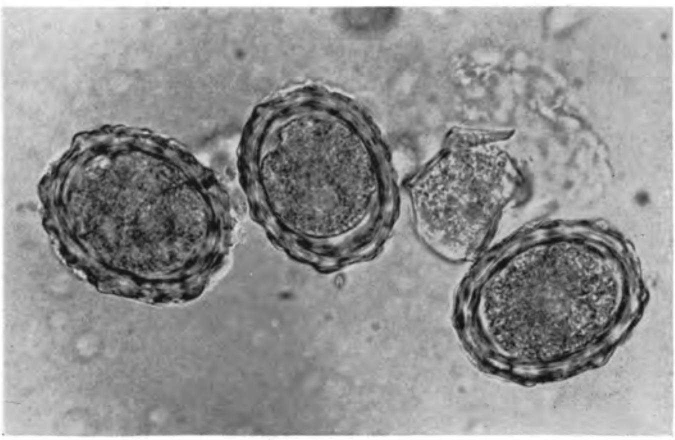

FIG. 71—Ova of **Ascaris lumbricoides.** Note the rough shell. The color is yellow. x 400.

SWINE

FIG. 72—Ova of **Macracanthorhynchus hirudinaceus,** the thorny-headed worm of swine. x 100.

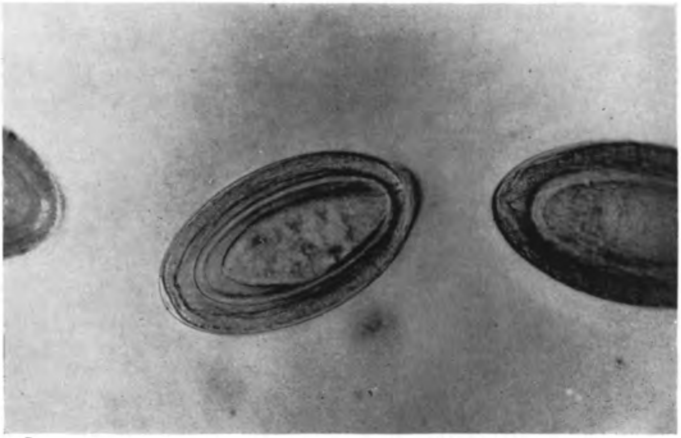

FIG. 73—Ova of **Macracanthorhynchus hirudinaceus.** The embryo is surrounded by three shells. The outer shell is dark brown. x 400.

SWINE

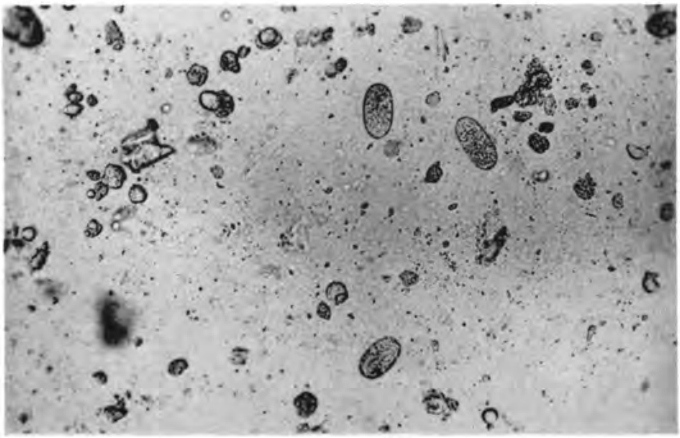

FIG. 74—Ova of **Oesophagostomum** sp., one of the four species of nodule worms of swine. x 100.

FIG. 75—Ova of **Oesophagostomum** sp. x 410.

SWINE

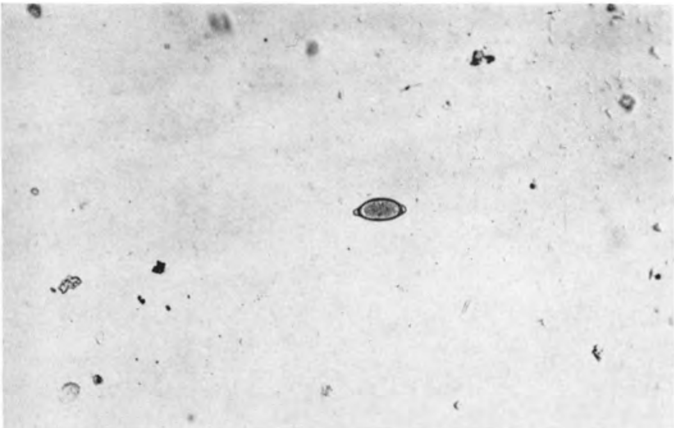

FIG. 76—Ovum of **Trichuris suis,** the whipworm of swine. x 100.

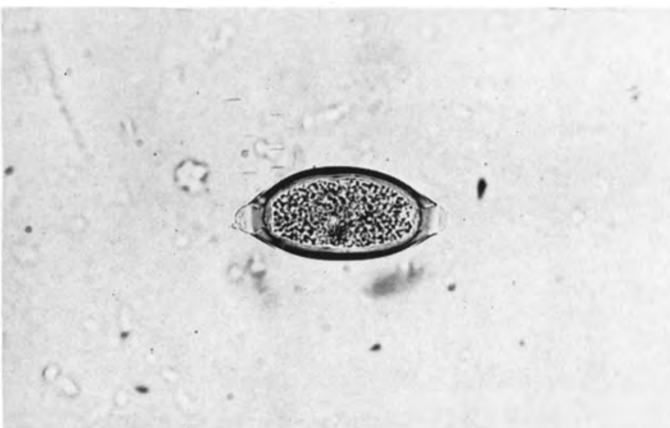

FIG. 77—Ovum of **Trichuris suis.** x 400.

SWINE

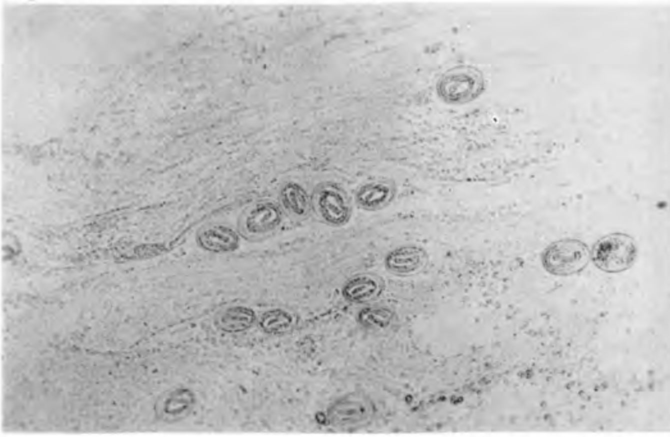

FIG. 78—Ova of **Metastrongylus apri,** one of the lungworms of swine. These were removed from the bronchial exudate but they may also be found in the feces. The eggs are embryonated when laid. x 100.

FIG. 79—Ova of **Metastrongylus apri.** x 400.

SWINE

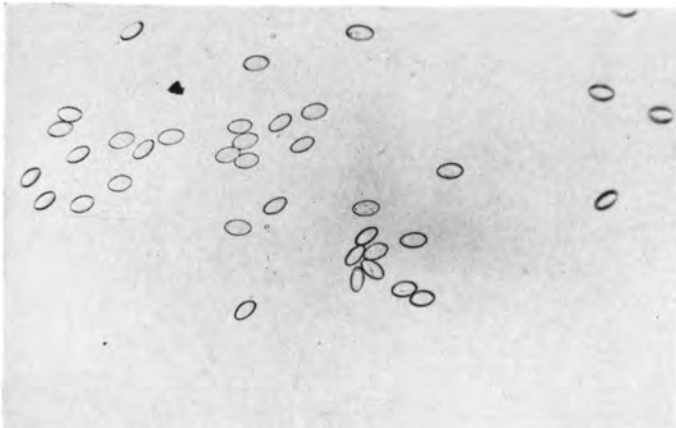

FIG. 80—Ova of **Ascarops strongylina,** one of the stomach worms of swine. x 100.

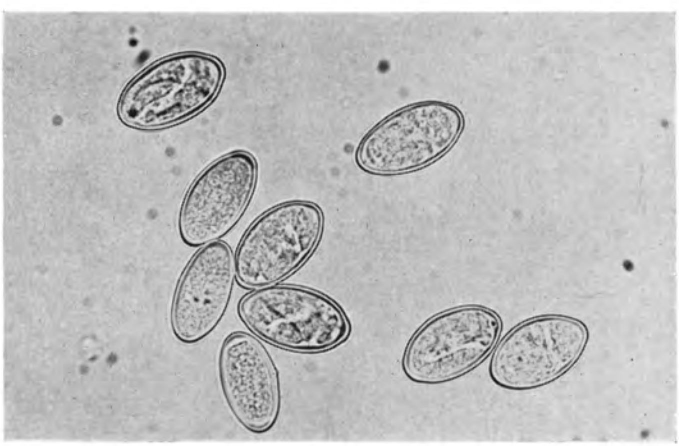

FIG. 81—Ova of **Ascarops strongylina.** x 410.

SWINE

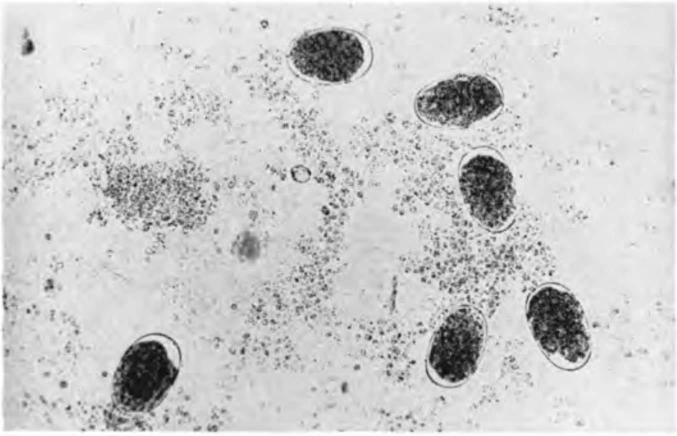

FIG. 82—Ova of **Stephanurus dentatus,** the kidney worm of swine. These eggs are found in urinary sediment and occasionally in the feces. x 100.

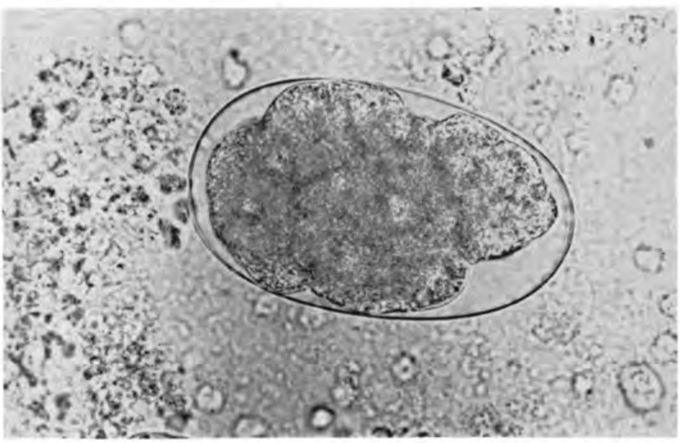

FIG. 83—Ovum of **Stephanurus dentatus.** x 410.

DOG, CAT, FOX

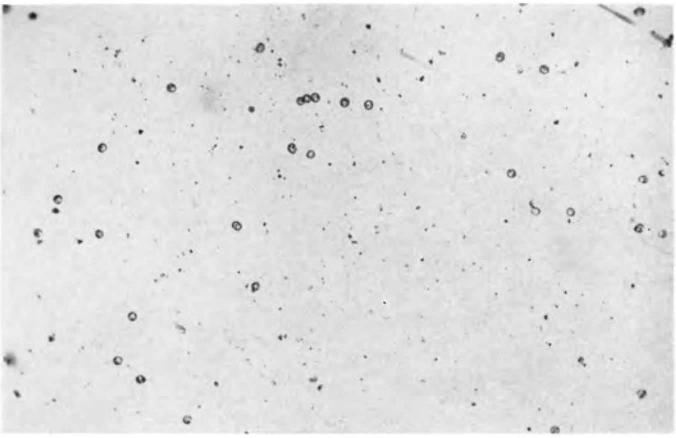

FIG. 84—Oocysts of **Isospora** sp., one of the coccidia of dogs, cats, and foxes. This is often referred to as the smaller form of **Isospora bigemina.** The oocysts are **not** sporulated when found in the feces. x 100.

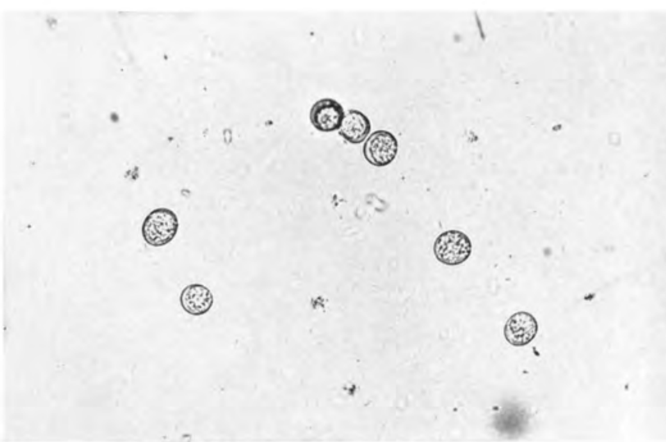

FIG. 85—Oocysts of **Isospora** sp. x 410.

DOG, CAT, FOX

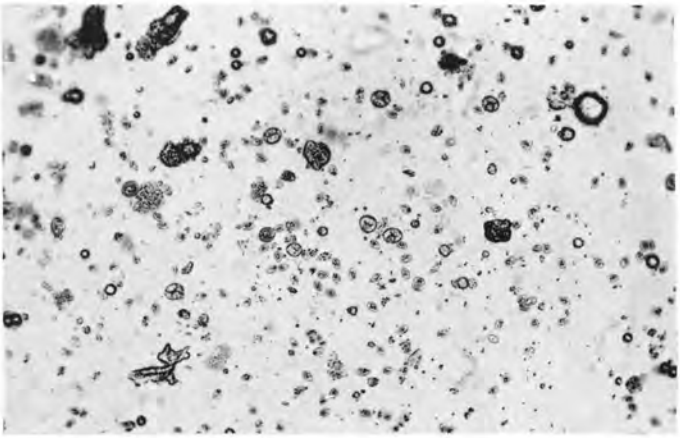

FIG. 86—Sporulated oocysts and sporocysts of **Isospora bigemina,** a coccidium of dogs, cats, and foxes. The larger oocysts seen are those of **Isospora rivolta.** x 100.

FIG. 87—Sporulated oocysts and sporocysts of **Isospora bigemina.** This coccidium is often referred to as the larger form of this species. The oocysts sporulate before leaving the body of the host and the delicate occyst wall frequently ruptures, liberating the two sporocysts, each of which contains four sporozoites. x 410.

DOG, CAT

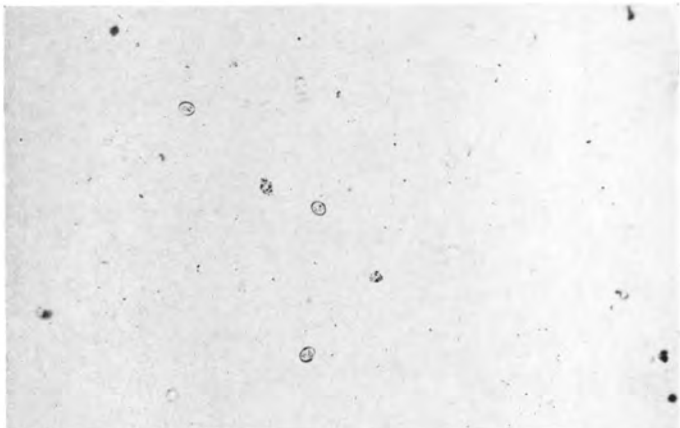

FIG. 88—Oocysts of **Isospora rivolta,** one of the coccidia of dogs and cats. These oocysts are intermediate in size between those of **I. bigemina** and **I. felis.** x 100.

FIG. 89—Oocyst of **Isospora rivolta.** x 410.

DOG, CAT

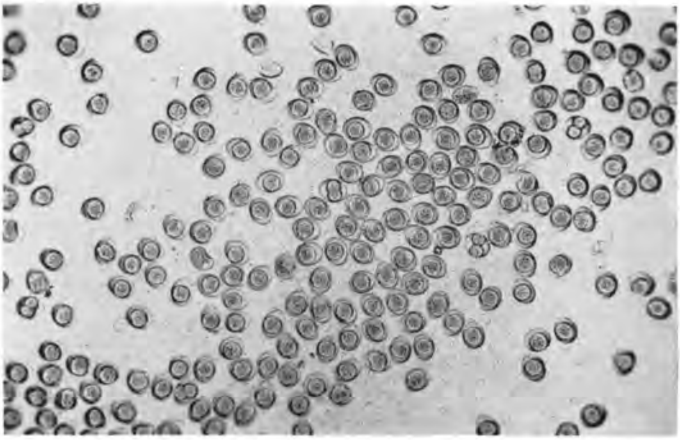

FIG. 90—Oocysts of **Isospora felis,** the largest species of the coccidia of dogs and cats. x 100.

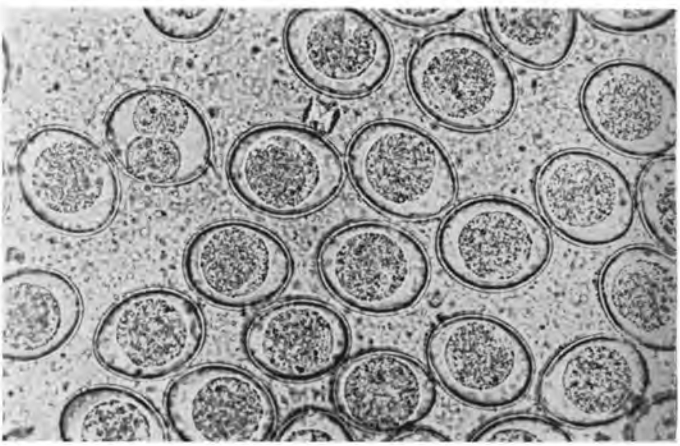

FIG. 91—Oocysts of **Isospora felis.** One shows beginning sporulation. x 410.

Note: A flagellate protozoon, **Giardia canis,** has been reported from the small intestine of dogs, and the same or a similar species from cats. Their morphology is similar to that of **Giardia bovis,** shown in Figs. 41, 42, 43, 44.

DOG, CAT, FOX, GOAT, SWINE, MINK, MUSKRAT, MAN

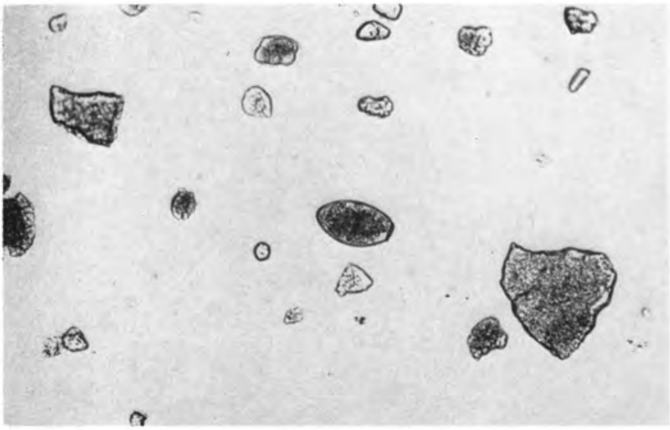

FIG. 92—Ovum of **Paragonimus westermanni,** the lung fluke of dogs, cats, foxes, goats, swine, mink, muskrat, and man. x 100.

FIG. 93—Ovum of **Paragonimus westermanni.** Note the prominent lid (operculum) at the right. x 410.

DOG, CAT, FOX, MAN

FIG. 94—Ova packets of **Dipylidium caninum**, the double-pored tapeworm of dogs, cats, foxes, and man. The smaller packets may be detected by flotation; the heavier packets sink in the centrifuge tube. x 100.

FIG. 95—An ova packet of **Dipylidium caninum**. Each egg in the packet is provided with six hooklets. x 400.

DOG, CAT, FOX

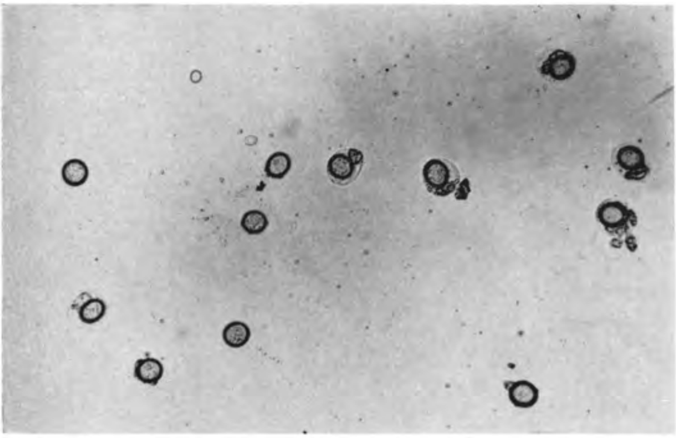

FIG. 96—Ova of **Taenia pisiformis,** one of the rabbit-cyst tapeworms of dogs, cats, and foxes. In general, tapeworm eggs leave the host in ripe tapeworm segments. However, eggs may be found by microscopic fecal examination. x 100.

FIG. 97—Ova of **Taenia pisiformis.** Note the radially striated shell and the embryonic hooklets. The egg at the right is contained within an embryonic membrane. x 400.

CAT, FOX

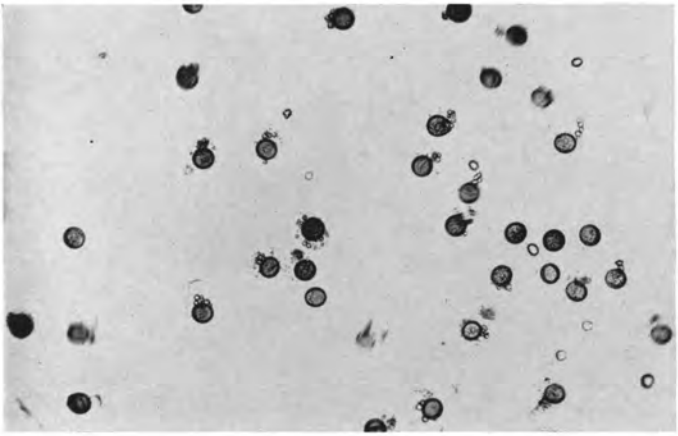

FIG. 98—Ova of **Taenia taeniaeformis,** a common tapeworm of cats and foxes. x 100.

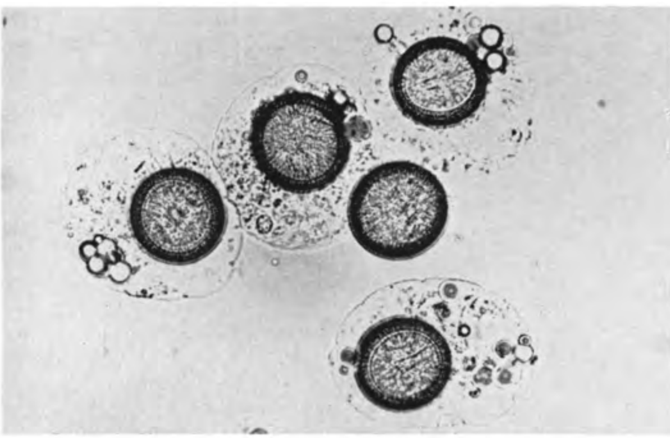

FIG. 99—Ova of **Taenia taeniaeformis.** Four of these are enclosed in embryonic membranes. x 410.

DOG, CAT, FOX, BEAR, MAN, OTHER FISH-EATING MAMMALS

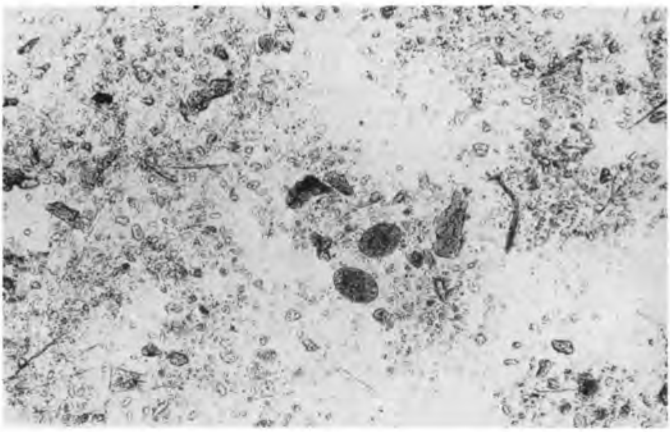

FIG. 100—Ova of **Diphyllobothrium latum,** the broad fish tapeworm of dogs, cats, foxes, bears, man, and other fish-eating mammals. x 100.

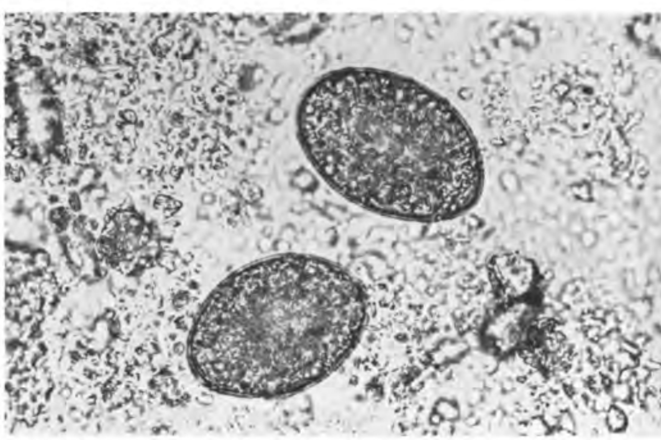

FIG. 101—Ova of **Diphyllobothrium latum.** x 410.

DOG, CAT, FOX

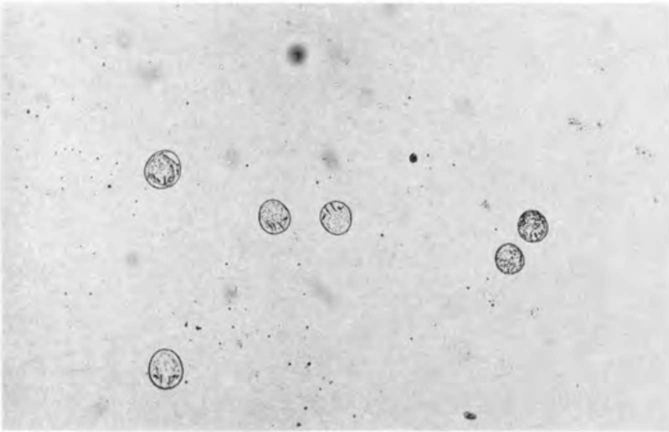

FIG. 102—Ova of **Mesocestoides variabilis,** a seldom-reported tapeworm of dogs, cats, and foxes. These eggs were removed from the egg-sac of a ripe segment. x 100.

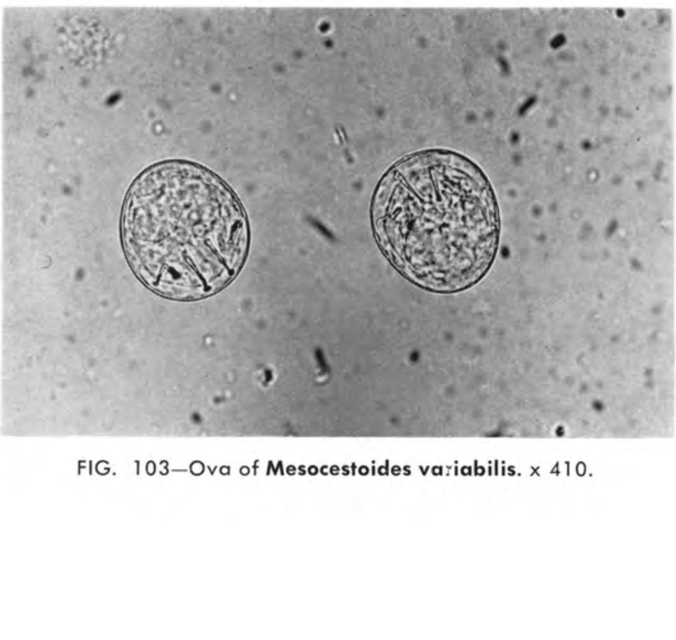

FIG. 103—Ova of **Mesocestoides variabilis.** x 410.

DOG, CAT, FOX

FIG. 104—Ova of **Ancylostoma caninum,** the commoner hookworm of dogs, cats, and foxes. x 100.

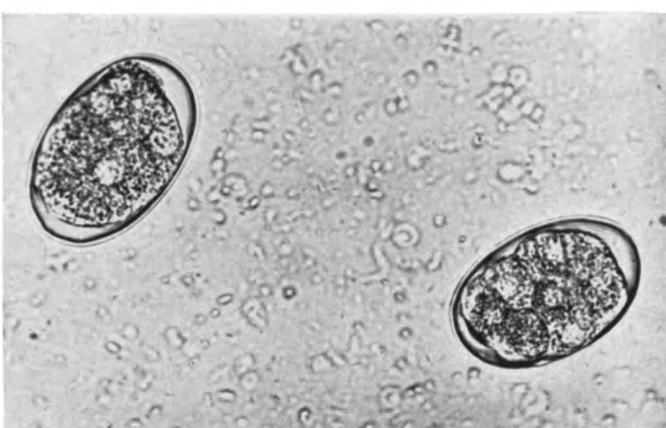

FIG. 105—Ova of **Ancylostoma caninum.** x 410.

DOG, CAT, FOX

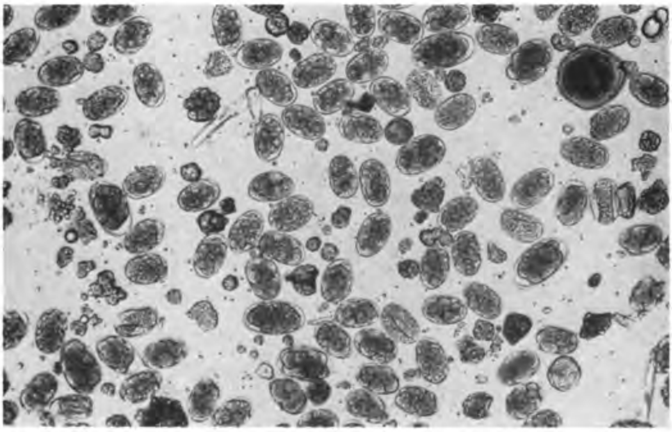

FIG. 106—Ova of **Uncinaria stenocephala** (larger ova) and **Ancylostoma caninum,** (smaller ova) hookworms of dogs, cats, and foxes. At the upper right is an ovum of **Toxocara canis,** one of the ascarids (see Figs. 108, 109). x 100.

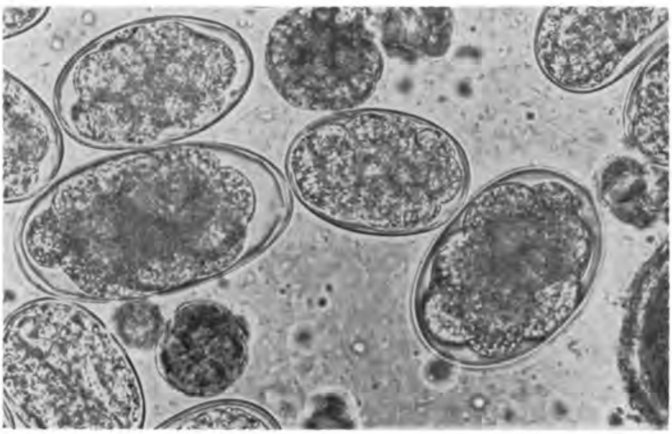

FIG. 107—Ova of **Uncinaria stenocephala** (larger ova) and **Ancylostoma caninum** (smaller ova). x 410.

DOG, CAT, FOX

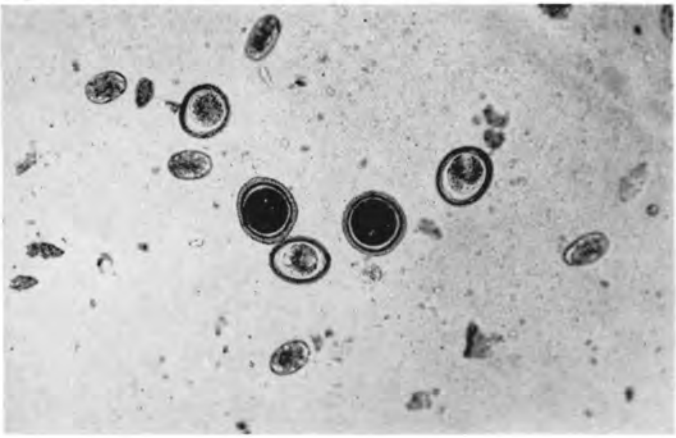

FIG. 108—Ova of **Toxocara canis** and **Toxascaris leonina,** both species of ascarids of dogs and foxes. The latter species also occurs in cats. Included are five ova of **Ancylostoma caninum,** the hookworm of dogs, cats, and foxes. x 100.

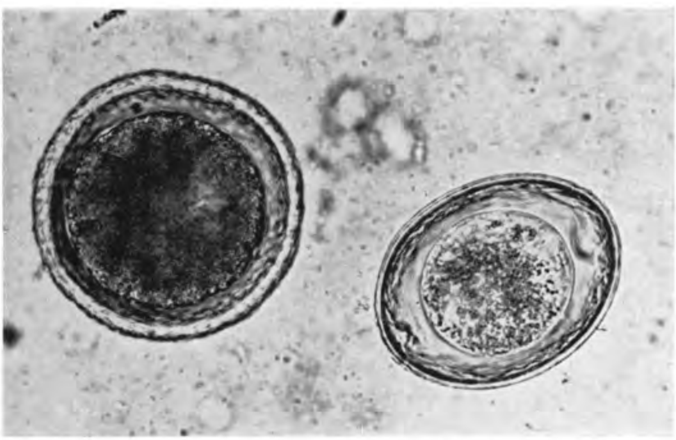

FIG. 109—Ova of **Toxocara canis (left)** and of **Toxascaris leonina (right).** The eggs of **Toxocara canis** are yellow. x 410.

CAT

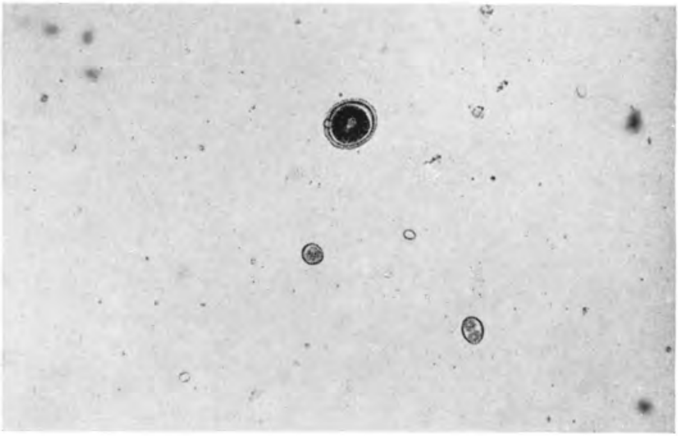

FIG. 110—Ovum of **Toxocara mystax,** an ascarid of cats. Also included are two oocysts of **Isospora felis.** x 100.

FIG. 111—Ovum of **Toxocara mystax** and an oocyst of **Isospora felis.** x 410.

DOG, CAT, FOX

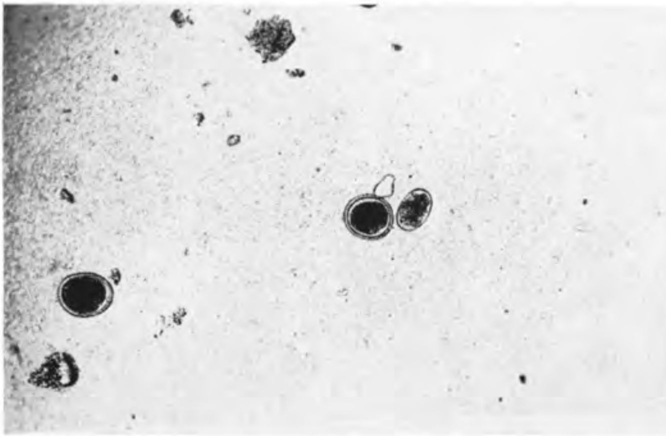

FIG. 112—Ova of **Toxocara mystax,** an ascarid of cats; and an ovum of **Ancylostoma caninum,** a hookworm of cats, dogs, and foxes. x 100.

FIG. 113—Ovum of **Toxocara mystax** and an ovum of **Ancylostoma caninum.** x 410.

DOG, FOX

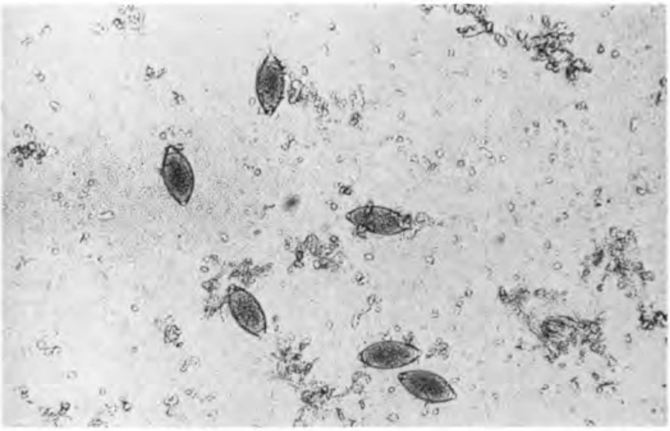

FIG. 114—Ova of **Trichuris vulpis,** the whipworm of dogs and foxes. x 100.

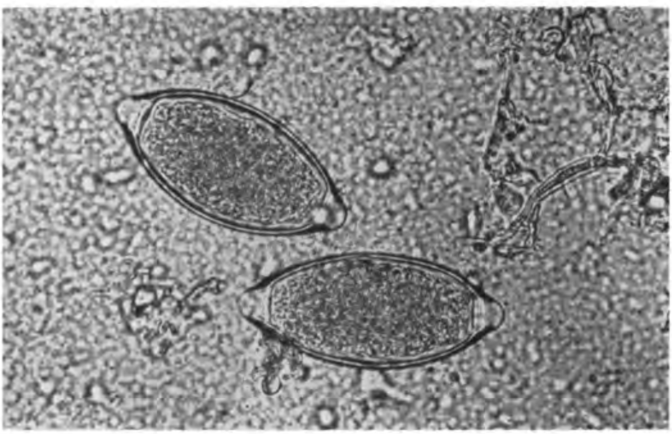

FIG. 115—Ova of **Trichuris vulpis.** Note the larger size and the smooth shell compared with lungworm ova (see Fig. 119). x 410.

DOG, CAT

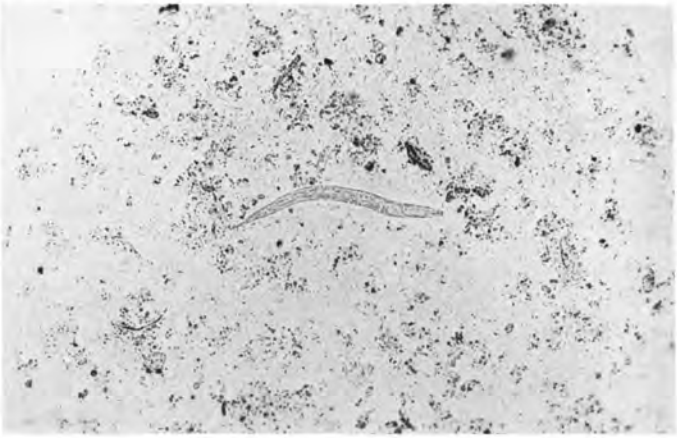

FIG. 116—Rhabditiform larva of **Strongyloides stercoralis,** the threadworm of dogs and cats. The ova hatch in the intestinal mucosa. x 100.

FIG. 117—Rhabditiform larva of **Strongyloides stercoralis.** x 410.

DOG, CAT, FOX

FIG. 118—Ova of **Capillaria aerophila,** the more common lungworm of dogs, cats, and foxes. x 100.

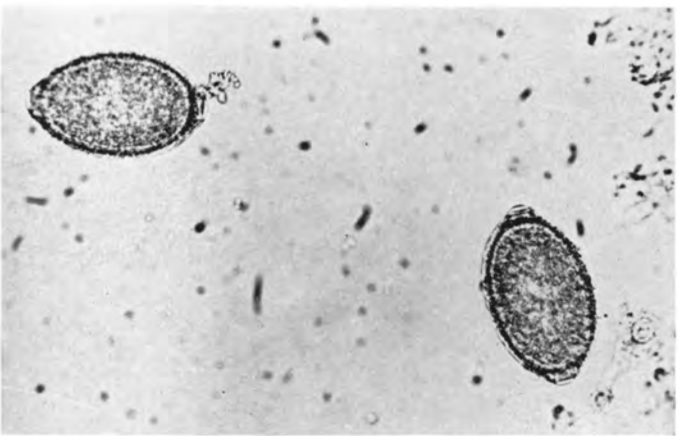

FIG. 119—Ova of **Capillaria aerophila.** The color is yellowish. The shells are finely granular and there is an operculum at each end. The size and the granular shell differentiate them from ova of **Trichuris vulpis,** the whipworm (Fig. 115). x 410.

DOG, FOX

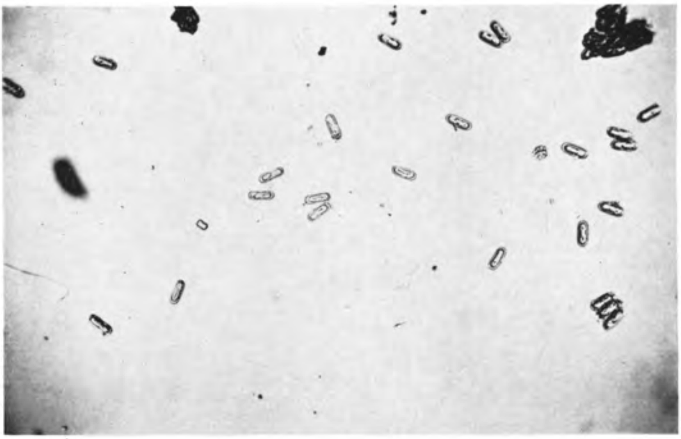

FIG. 120—Ova of **Spirocerca lupi,** the esophageal worm of dogs and foxes. x 100.

FIG. 121—Ova of **Spirocerca lupi.** These eggs are embryonated when laid. x 410.

DOG, CAT, FOX

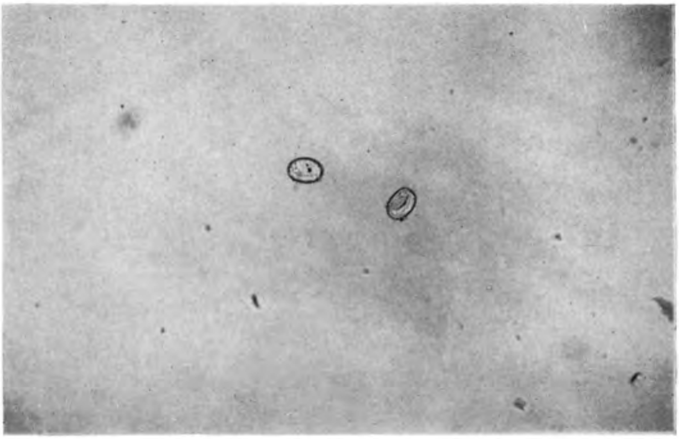

FIG. 122—Ova of **Physaloptera rara,** a stomach worm of dogs, cats, and foxes. x 100.

FIG. 123—Ova of **Physaloptera rara.** These eggs are embryonated when laid. x 410.

DOG, CAT, FOX

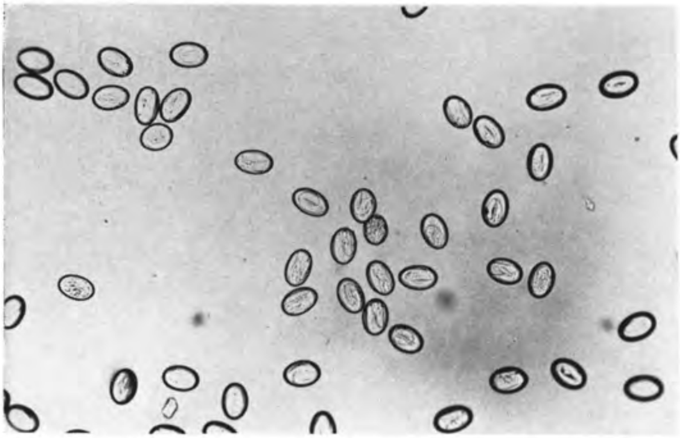

FIG. 124—Ova of **Physaloptera praeputialis,** a stomach worm of dogs, cats, and foxes. x 100.

FIG. 125—Ova of **Physaloptera praeputialis.** These eggs are embryonated when laid. x 410.

DOG, FOX

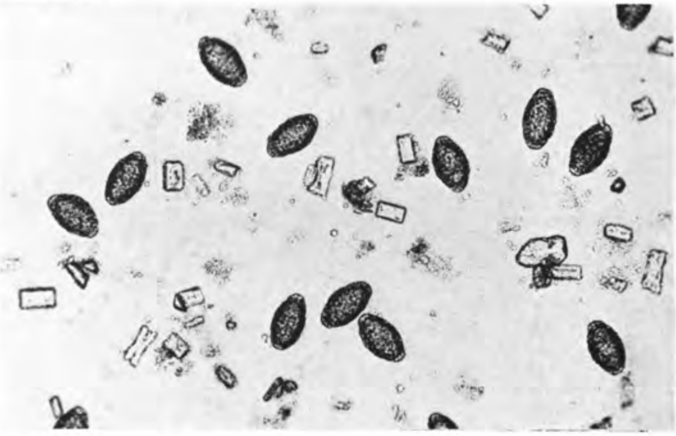

FIG. 126—Ova of **Dioctophyma renale,** the giant kidney worm of dogs and foxes. These eggs are usually found in urinary sediment (note triple phosphate crystals). x 100.

FIG. 127—Ova of **Dioctophyma renale.** The shells are thick and rough. The color is yellowish-brown. x 410.

DOG

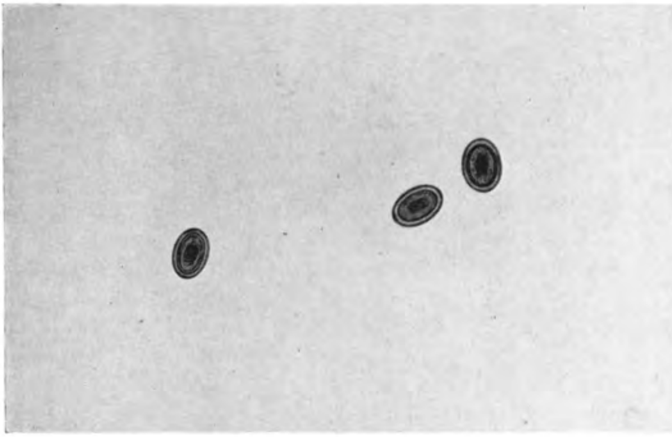

FIG. 128—Ova of **Oncicola canis,** the thorny-headed worm of dogs. x 100.

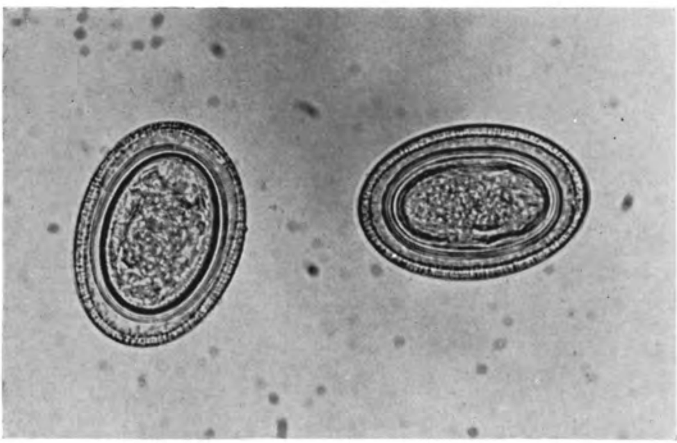

FIG. 129—Ova of **Oncicola canis.** Note the three shells enclosing the embryo. x 410.

DOG

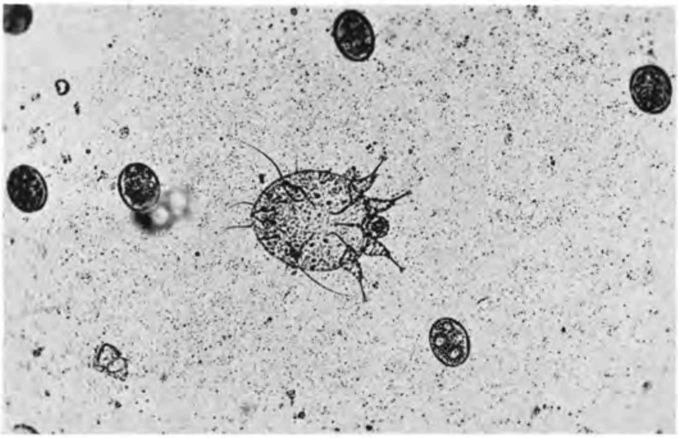

FIG. 130—A larva of **Sarcoptes scabiei** var. **canis**, the sarcoptic mange mite of dogs; also several ova of **Ancylostoma caninum**, a hookworm, in dog feces. Mange, especially in dogs and cats, may be diagnosed by fecal examination if the host happens to ingest mites when biting the skin lesions. x 100.

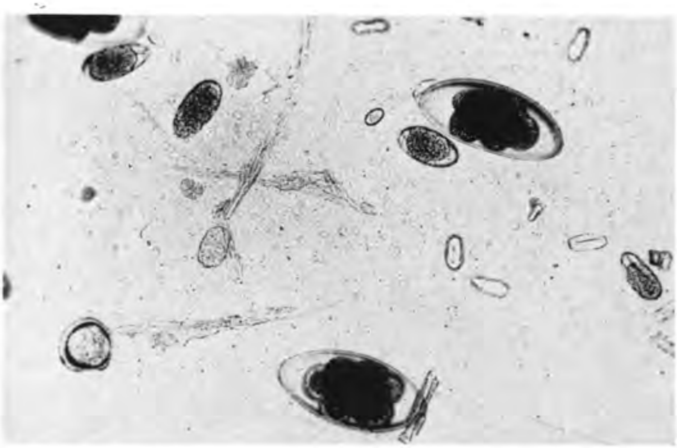

FIG. 131—Spurious parasites. The feces of this dog contains ova and oocysts of sheep parasites. The dog's food was contaminated by sheep feces. The field contains ova of **Nematodirus spathiger, Moniezia expansa, Strongyloides papillosus,** also an unidentified nematode ovum and a coccidial oocyst. x 100.

DOG

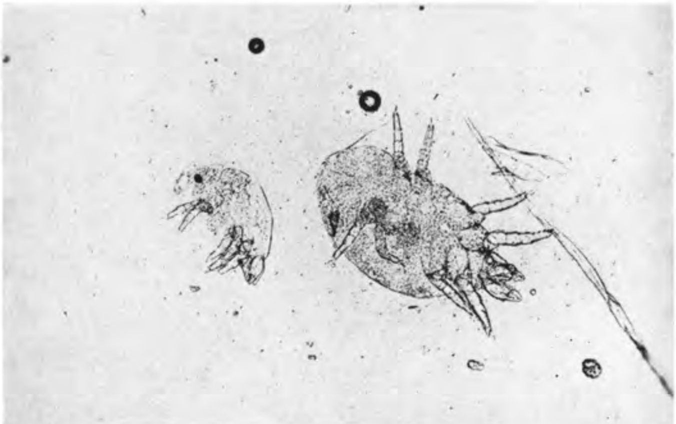

FIG. 132—Pseudoparasite. An adult and a larval "grain" mite in the feces of a dog. x 100.

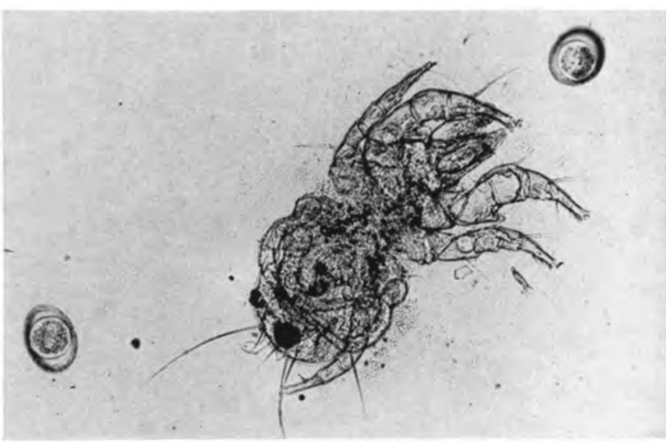

FIG. 133—Pseudoparasite. An adult "grain" mite and two ova of **Toxocara canis** appear in this sample of dog feces. x 100.

DOG

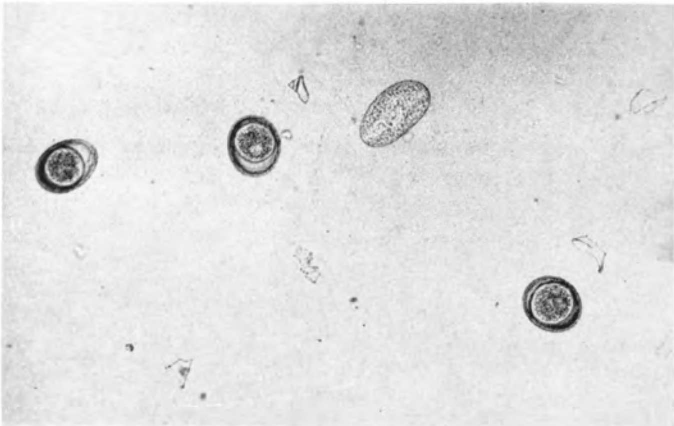

FIG. 134—Pseudoparasite. An ovum of a "grain" mite and three ova of **Toxocara canis** appear in this sample of dog feces. x 100.

DOG

FIG. 135—Pseudoparasite. Pine pollen in dog feces. The color is pale brown. x 100.

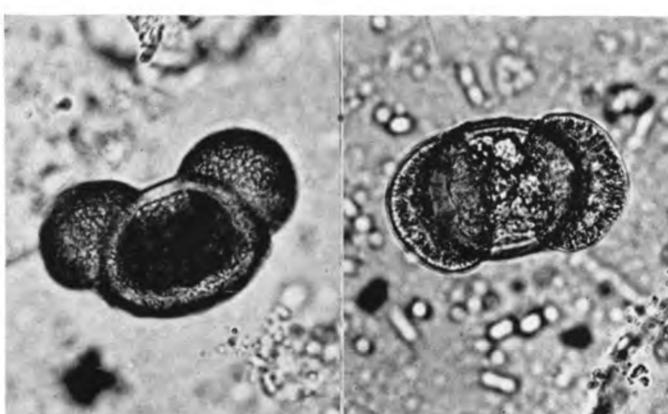

FIG. 136—Pine pollen in the feces of a dog. Side view of a pollen grain **(left)**, showing the two wing-like floats. View from above at the right. x 410.

DOG

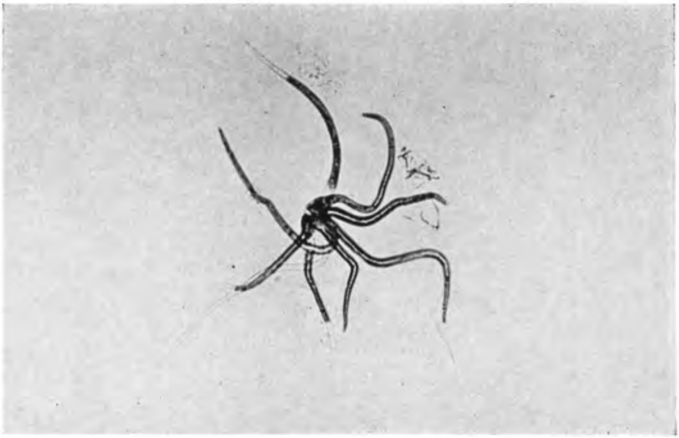

FIG. 137—Pseudoparasite. Plant hairs from dog feces. These resemble the groups of hair-like projections seen on the under surface of oak leaves. x 100.

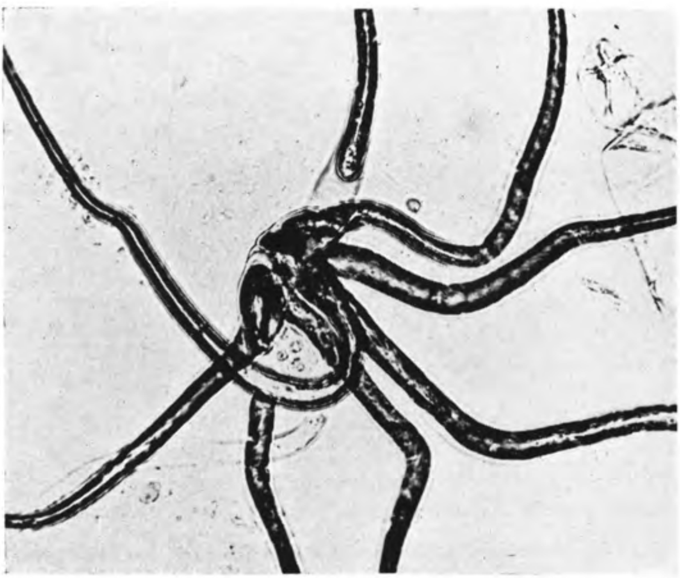

FIG. 138—Plant hairs from dog feces. x 338.

DOG

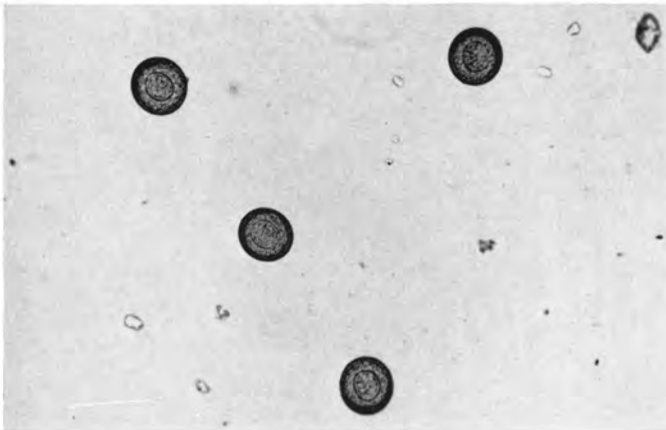

FIG. 139—Spurious parasite. The feces of this dog contains ova of **Hymenolepis diminuta,** a tapeworm of rats, mice, and man. Presumably the dog ingested the small intestine of an infected rodent. These eggs are yellow in color. x 100.

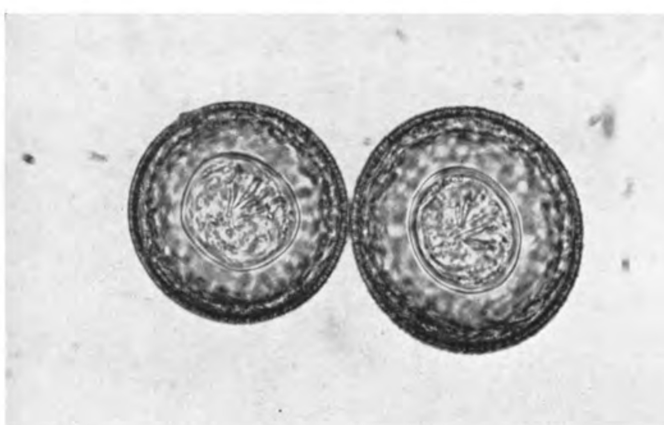

FIG. 140—**Hymenolepis diminuta** ova in dog feces. Note the six hooklets in each embryo. x 410.

DOG

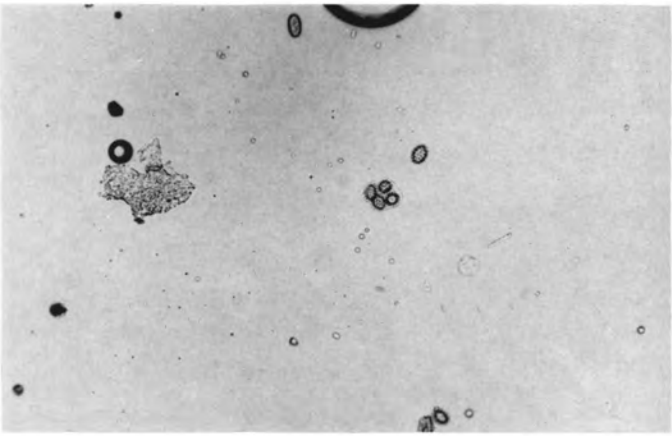

FIG. 141—Pseudoparasite. Corn smut spores in the feces of a dog. These resemble certain tapeworm ova under low power. x 100.

FIG. 142—Corn smut spores in feces. Note the spiny covering. x 410.

DOG

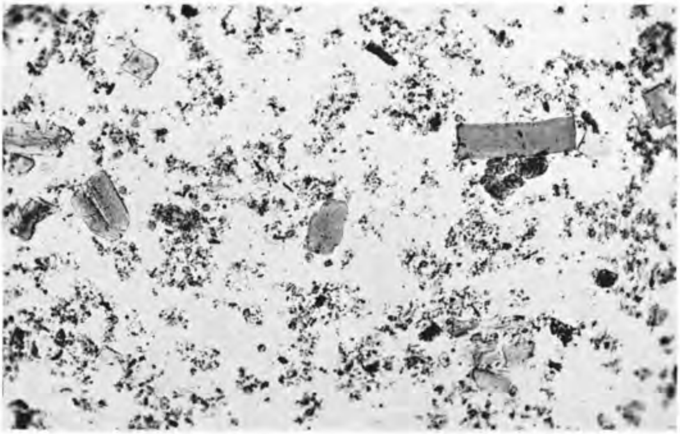

FIG. 143—Undigested muscle in a dog's feces. x 100.

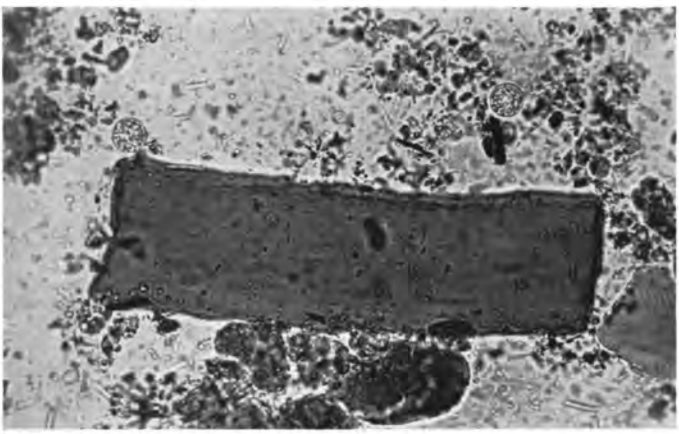

FIG. 144—Undigested muscle in a dog's feces. x 410.

CHICKEN

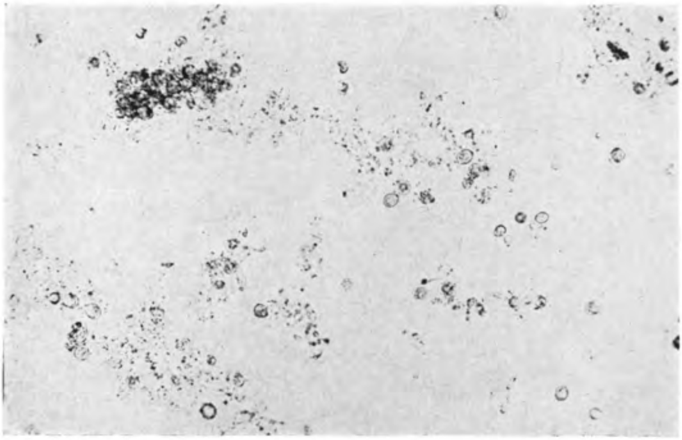

FIG. 145—Oocysts of **Eimeria tenella,** the cecal coccidium of chickens. x 100.

FIG. 146—Oocysts of **Eimeria tenella.** x 410.

TURKEY

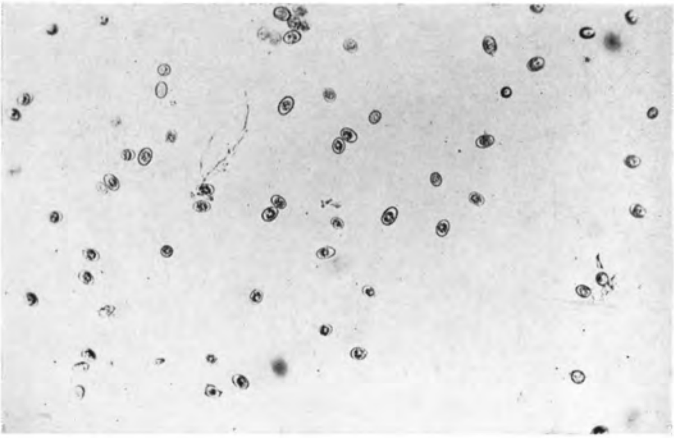

FIG. 147—Oocysts of **Eimeria meleagridis** and **Eimeria meleagrimitis,** two species of coccidia of turkeys. x 100.

FIG. 148—Two oocysts of **Eimeria meleagridis** and four oocysts of **Eimeria meleagrimitis.** x 410.

PHEASANT, TURKEY

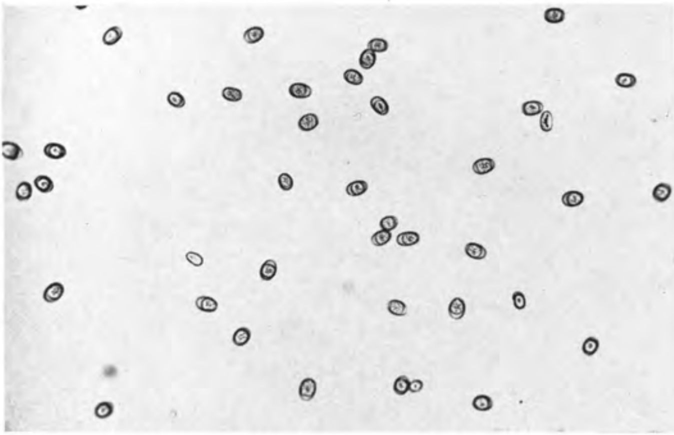

FIG. 149—Oocysts of **Eimeria dispersa** and **Eimeria phasiani,** coccidia of pheasants. **Eimeria dispersa** is also a coccidium of turkeys. x 100.

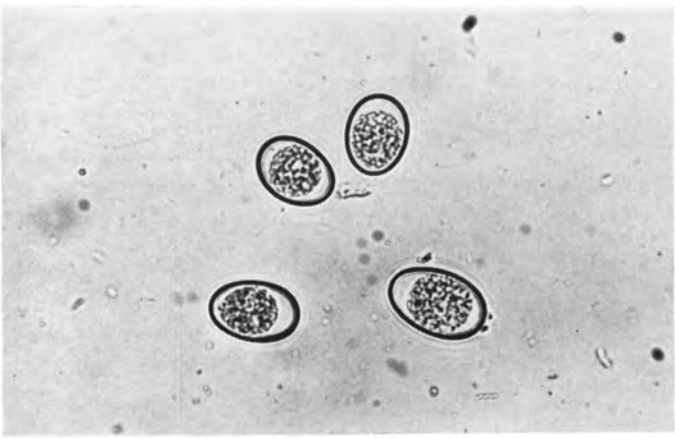

FIG. 150—Oocysts of **Eimeria dispersa** and **Eimeria phasiani.** The latter species is slightly the larger. x 410.

PIGEON

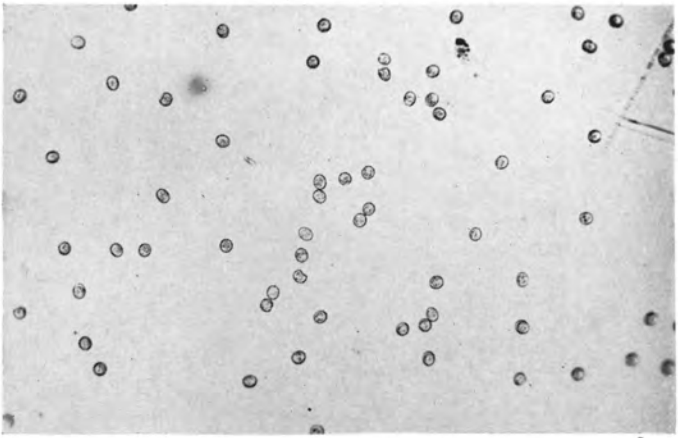

FIG. 151—Oocysts of **Eimeria labbeana,** the coccidium of pigeons. x 100.

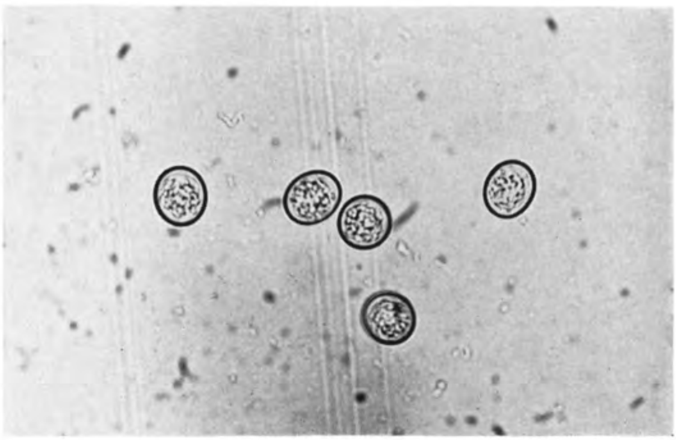

FIG. 152—Oocysts of **Eimeria labbeana.** x 410.

CHICKEN, TURKEY

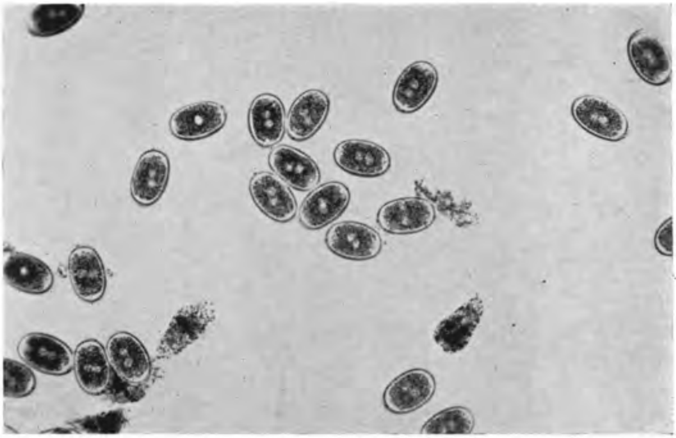

FIG. 153—Ova of **Ascaridia galli,** the ascarid of the chicken and rarely of the turkey. x 100.

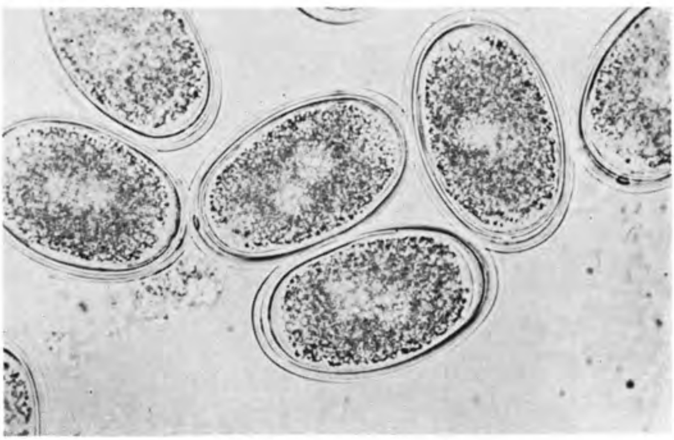

FIG. 154—Ova of **Ascaridia galli.** x 400.

CHICKEN, TURKEY, GUINEA FOWL, QUAIL, PHEASANT

FIG. 155—Ova of **Heterakis gallinae,** the cecal worm of chickens, turkeys, guinea fowl, quail, and pheasants. x 100.

FIG. 156—Ova of **Heterakis gallinae.** x 410.

TURKEY, DUCK, QUAIL, PHEASANT

FIG. 157—Ova of **Capillaria contorta,** the crop capillarid of turkeys, ducks, quail, and pheasants. x 100.

FIG. 158—Ova of **Capillaria contorta.** There is an operculum at each pole. x 410.

CHICKEN, TURKEY, PHEASANT

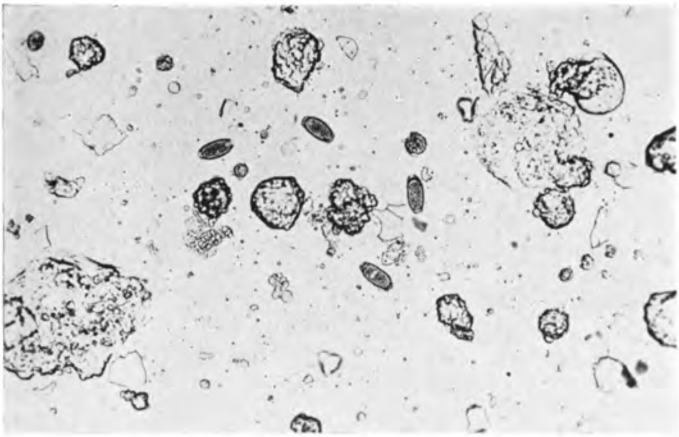

FIG. 159—Ova of **Capillaria caudinflata,** a capillarid worm of the small intestine of chickens, turkeys, and pheasants. x 100.

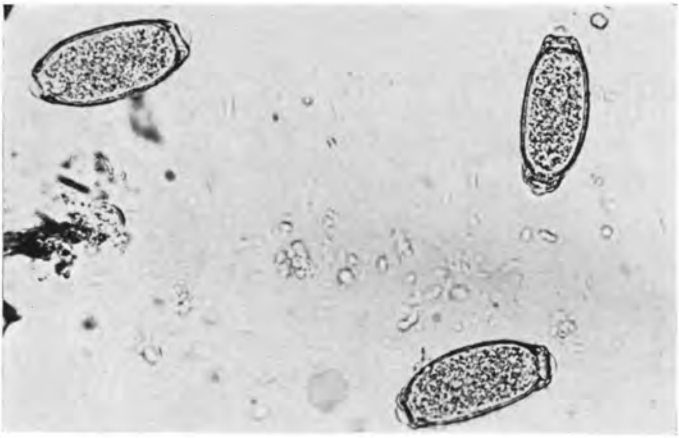

FIG. 160—Ova of **Capillaria caudinflata.** Note the operculum at each pole. x 410.

POULTRY

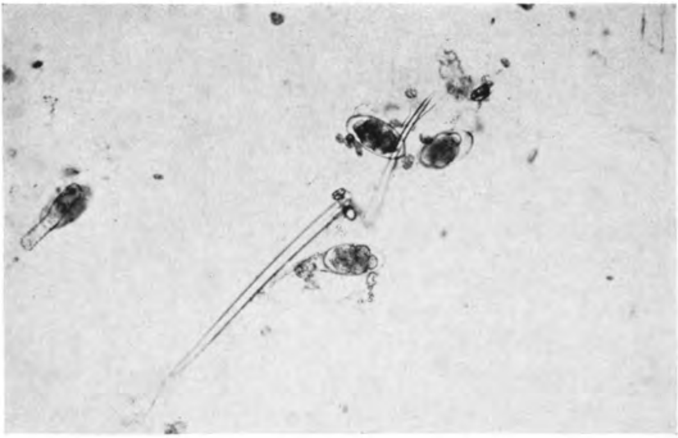

FIG. 161—Ova of **Syngamus trachea,** the gapeworm of poultry. x 100.

FIG. 162—Ovum of **Syngamus trachea.** There is an operculum at both of the poles. x 410.

CHICKEN

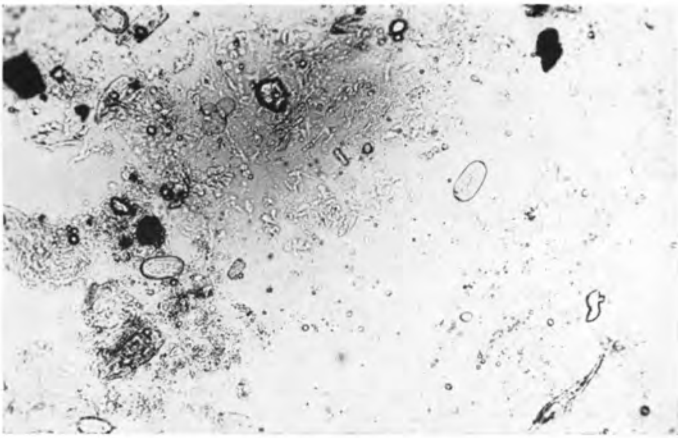

FIG. 163—Ova of **Tetrameres americana**, the globular stomach worm of chickens. x 100.

FIG. 164—Ovum of **Tetrameres americana**. The eggs of this nematode are embryonated when laid. x 410.

CHICKEN, TURKEY, GUINEA FOWL, PIGEON

FIG. 165—Ova of **Dispharynx nasuta,** the spiral stomach worm of chickens, turkeys, guinea fowl, and pigeons. x 100.

FIG. 166—Ova of **Dispharynx nasuta.** The ova are embryonated when laid. x 410.

RABBIT, HARE

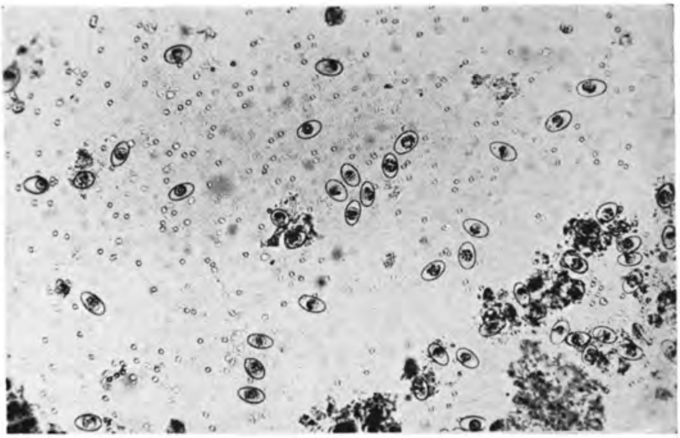

FIG. 167—Oocysts of **Eimeria stiedae,** the hepatic coccidium of rabbits and hares. These were removed from the bile duct. They may also be found in the feces. x 100.

FIG. 168—Oocysts of **Eimeria stiedae.** x 410.

RABBIT

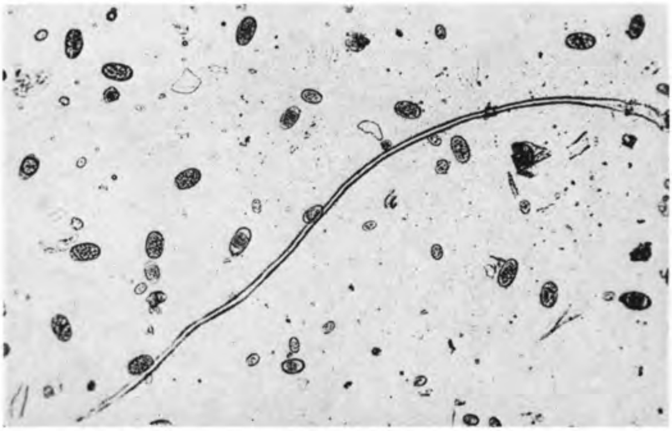

FIG. 169—Oocysts of several **Eimeria** species, intestinal coccidia of rabbits. A long plant hair is present. x 100.

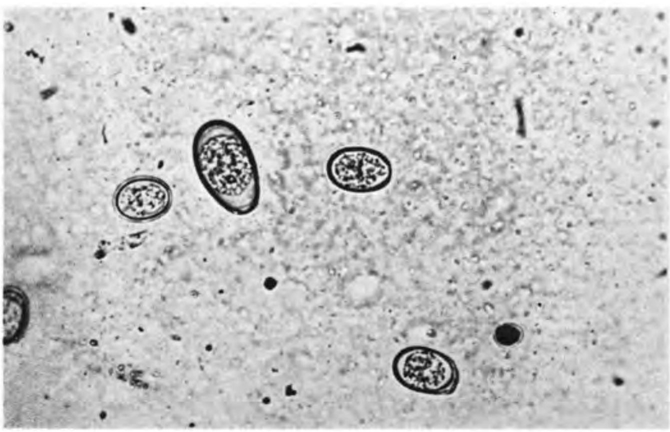

FIG. 170—Oocysts of three **Eimeria** species, coccidia of rabbits. x 410.

RABBIT, GUINEA PIG

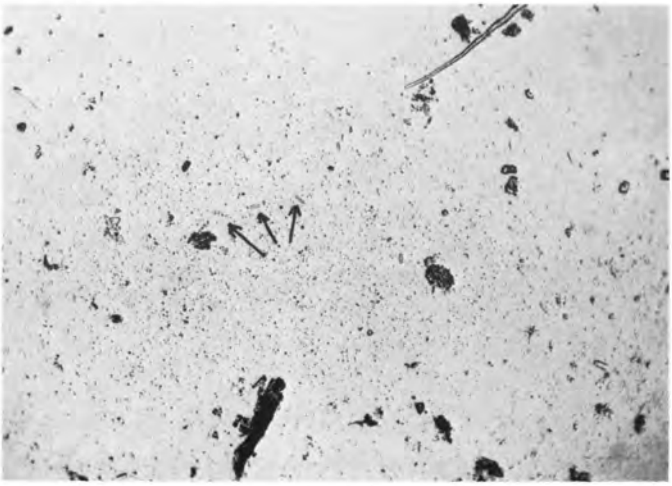

FIG. 171—Pseudoparasite. **Saccharomycopsis guttulatus,** a yeast commonly found in the feces of rabbits and guinea pigs. It is not believed to be pathogenic. Arrows point to the yeasts. x 100.

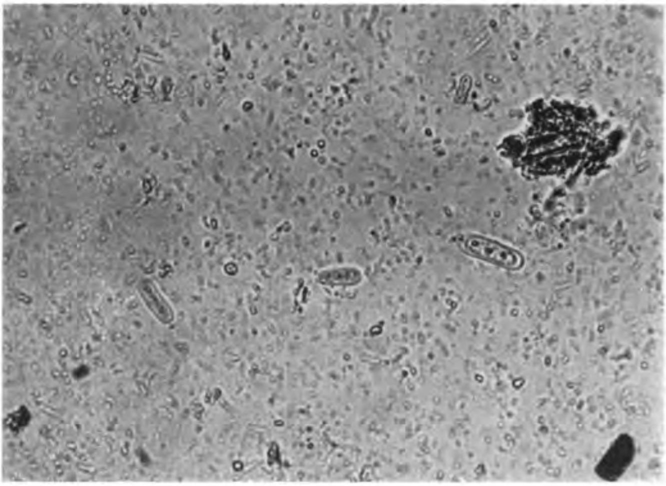

FIG. 172—Pseudoparasite. **Saccharomycopsis guttulatus.** x 410.

MAN

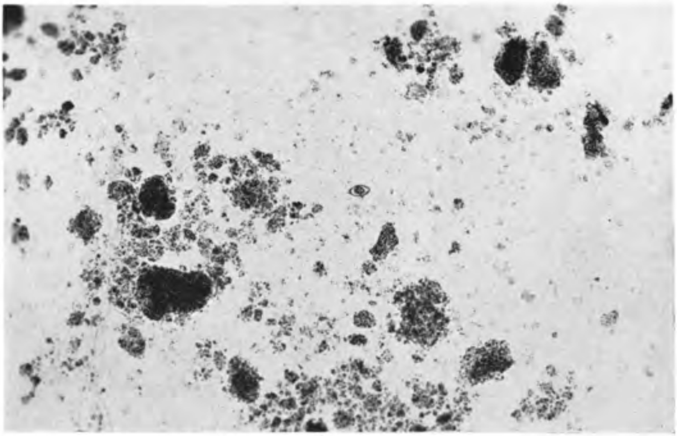

FIG. 173—Oocyst of **Isospora hominis,** the coccidium reported as occurring in man. From human feces. x 100.

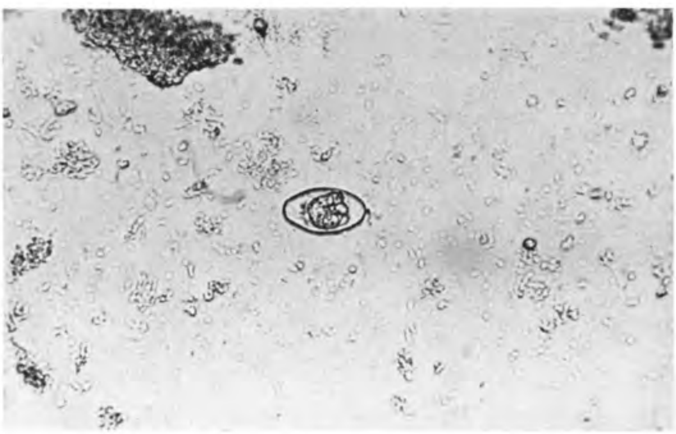

FIG. 174—Oocyst of **Isospora hominis.** x 410.

MAN

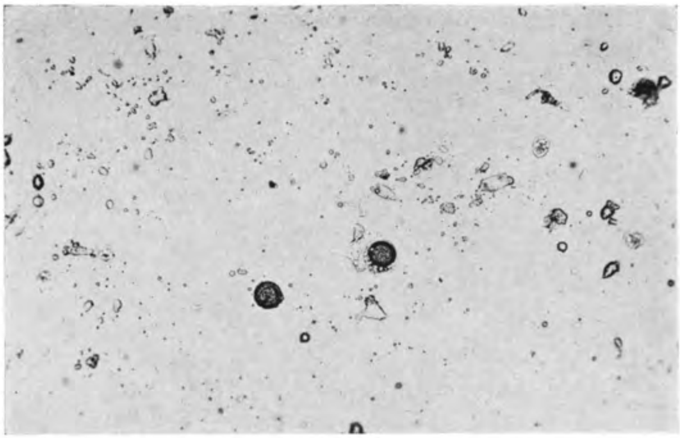

FIG. 175—Ova of **Taenia saginata,** the beef tapeworm of man. x 100.

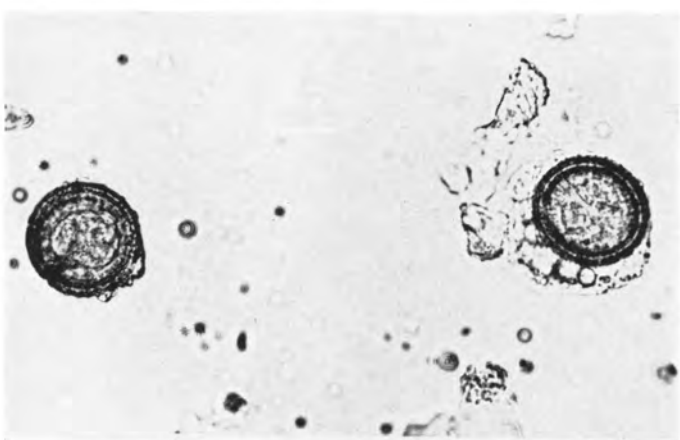

FIG. 176—Ova of **Taenia saginata.** The egg at the right is contained within an embryonic membrane. x 410.

MAN, RAT, MOUSE

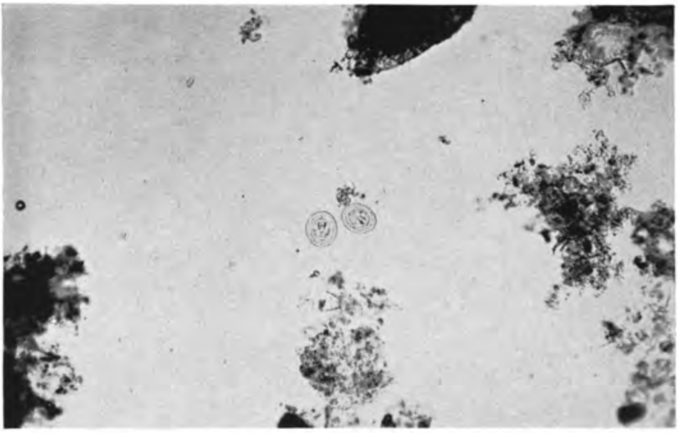

FIG. 177—Ova of **Hymenolepis nana,** the dwarf tapeworm of man, rats, and mice. x 100.

FIG. 178—Ova of **Hymenolepis nana**. There are from four to eight slender filaments on each polar thickening of the inner shell membrane. x 385.

MAN

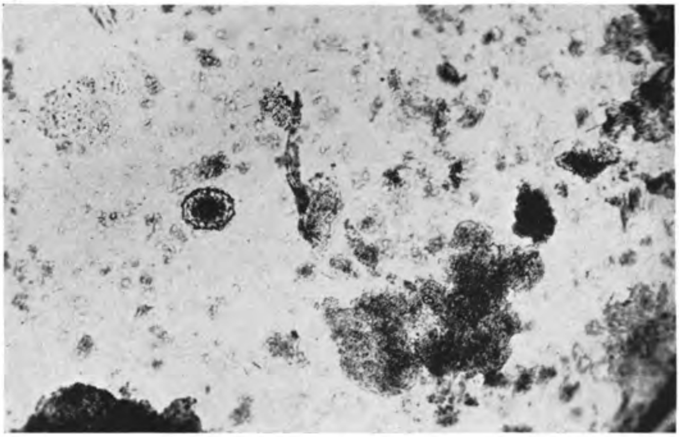

FIG. 179—Ovum of **Ascaris lumbricoides,** the ascarid of man. x 100.

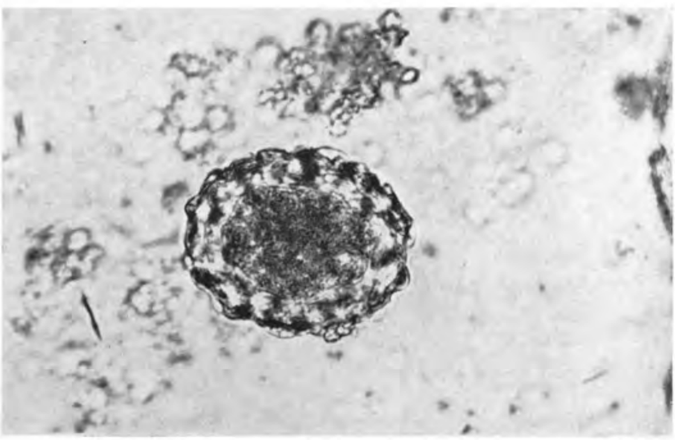

FIG. 180—Ovum of **Ascaris lumbricoides.** x 410.

MAN

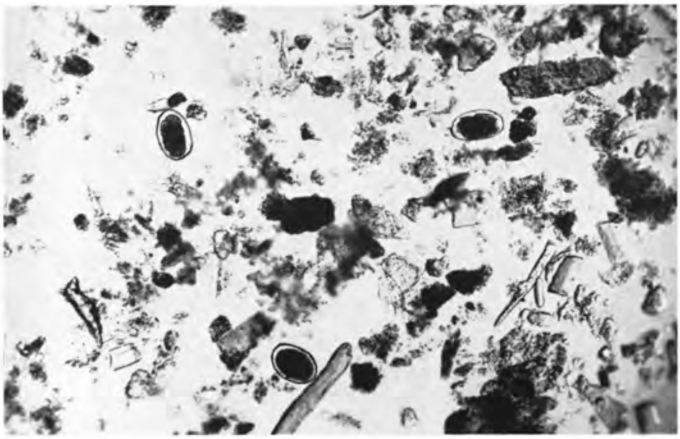

FIG. 181—Ova of **Necator americanus,** the new-world hookworm of man. Simple smear. x 100.

FIG. 182—Ovum of **Necator americanus.** x 410.

MAN

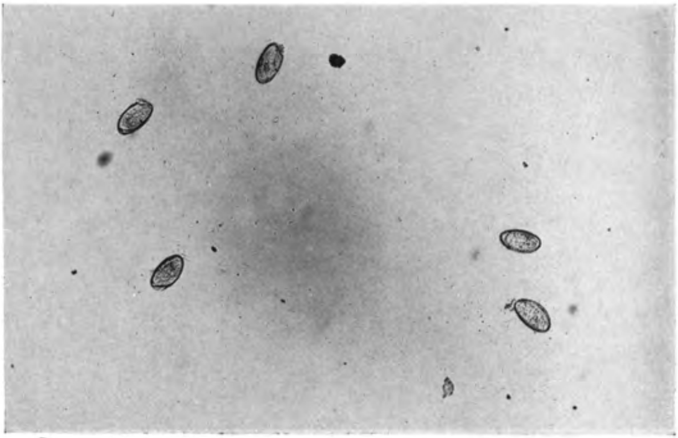

FIG. 183—Ova of **Enterobius vermicularis,** the pinworm or rectal worm of man. x 100.

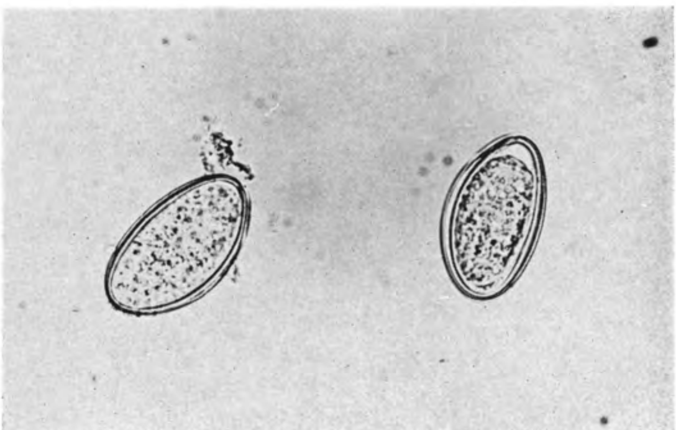

FIG. 184—Ova of **Enterobius vermicularis.** x 410.

MAN

FIG. 185—Larvae of **Strongyloides stercoralis,** the threadworm of man. x 100.

FIG. 186—Larva of **Strongyloides stercoralis.** x 410.

MAN

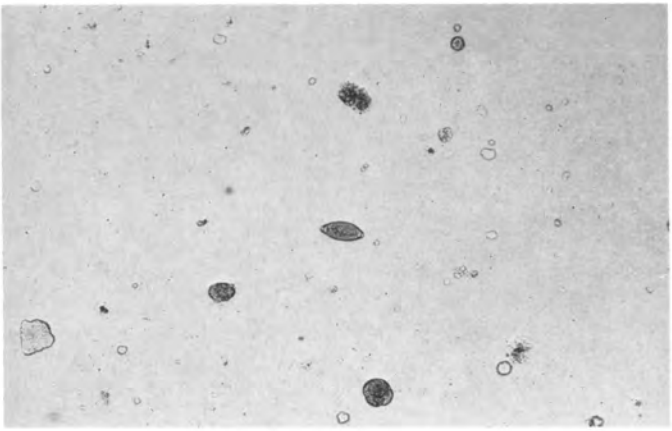

FIG. 187—Ovum of **Trichuris trichiura,** the whipworm of man. x 100.

FIG. 188—Ovum of **Trichuris trichiura** of man. Note the resemblance to the ova of the swine whipworm (Fig. 77). x 410.

MAN

FIG. 189—Pseudoparasite. Banana seeds in human feces. Grossly these resemble small brownish tapeworm segments. x 3.

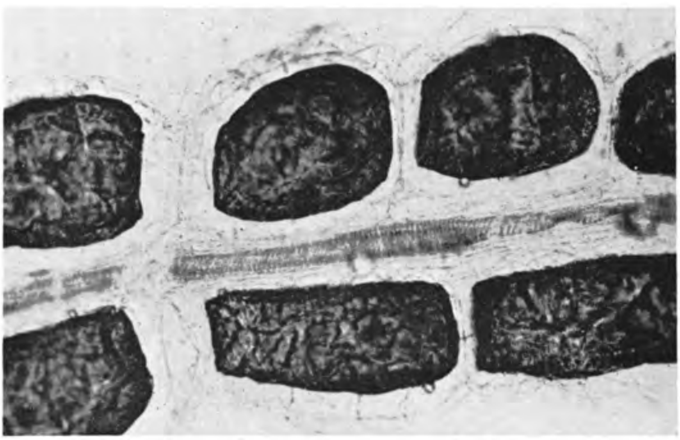

FIG. 190—Banana seeds in human feces. x 100.

SECTION 2

Examination for Mites of the Skin and of the Internal Organs

MORE THAN 50 species of mites have been reported to live on or in domesticated mammals and birds of North America. These include the parasitic mange and scab mites, scaly-leg mite, depluming mite, ear mites, feather and quill mites, flesh mite, air-sac mite, chigger mites, roost mite, sinus mite, and nasal mites.

For a discussion of parasitic (and nonparasitic) mites, reference is made to the book by Baker and Wharton (1952): *An Introduction to Acarology*.

The mites (also the ticks) belong in the phylum Arthropoda (animals with an exoskeleton and jointed limbs). Arthropods without antennae and mandibles belong in the class Arachnida (spider-like animals). In the class Arachnida is the order Acarina, which includes the mites and the ticks; this order comprises "arachnides with mouthparts set off from the rest of the body on a false head (capitulum or gnathosoma)" and in which body segmentation is greatly reduced or absent.

Mites are smaller than ticks, most species being either microscopic or under 1 mm. in length. They are covered by a relatively soft, often translucent skin through which respiration takes place, in the smaller species. The larger species breathe through skin openings (stigmata) connected with tracheal tubes.

The body may be ornamented by spines or hairs (setae), or by scale-like plates. The legs (4 pairs for adults and nymphs; 3 pairs for larvae) are provided with claw-like hooks or suctorial cups (Figs. 227, 235).

Depending upon the species, the food of parasitic mites includes mainly blood, lymph, living and dead epithelial cells, or feathers. Mouthparts are adapted for either piercing or chewing.

The mite life cycle usually begins with the laying of the egg, from which a six-legged larva emerges. After feeding, the skin is shed and the eight-legged but sexually immature nymph appears.

Following one or more skin molts, the sexually mature adult mite is formed. Variations occur in the life cycle of certain mite species. For example, the air-sac mite, *Cytodites nudus* of poultry is viviparous; the sinus mite, *Pneumonyssus caninum* of dogs has not been observed to have a nymph stage.

According to Baker and Wharton (1952) the parasitic mites are grouped under three suborders:

I. Suborder SARCOPTIFORMES (7 families)
II. Suborder TROMBIDIFORMES (5 families)
III. Suborder MESOSTIGMATA (4 families)

Following is a brief listing and description of the parasitic mites of domesticated animals; by suborders and families.

I. Suborder SARCOPTIFORMES

Family 1. SARCOPTIDAE

Three important genera of mange and scab mites belong in this family, namely the genera (1) Sarcoptes, (2) Notoedres, and (3) Cnemidocoptes.

(1) *Sarcoptic mange mites.* These mites are the cause of sarcoptic mange or itch. The fertilized females work their way deeply into the epidermis, forming tunnels where they deposit their eggs. Close proximity to nerve endings results in intense irritation. The skin thickens and rather dense crusts form (Fig. 208). The infestation usually involves thin-skinned areas first. There is considerable loss of hair. These mites cause the most common form of mange in swine and horses. The morphologic characteristics of sarcoptic mites are shown in Table 1, page 128, and Fig. 201.

Species and hosts:

Sarcoptes scabiei var. *equi* — Horse
Sarcoptes scabiei var. *bovis* — Cattle (Fig. 207)
Sarcoptes scabiei var. *ovis* — Sheep
Sarcoptes scabiei var. *caprae* — Goat
Sarcoptes scabiei var. *suis* — Swine (Figs. 209 to 213)
Sarcoptes scabiei var. *canis* — Dog (Figs. 214 to 218)
Sarcoptes scabiei var. *vulpis* — Fox

(2) *Notoedric mange mites.* These resemble the sarcoptic mites but they are somewhat smaller; and the anus is located on the dorsal abdominal area rather than terminally (Fig. 221). Notoedric mange is fairly common on cats and rabbits. Lesions are first noticed on the face and other areas of the head, later spreading to various parts of the body, particularly to the forelegs. Advanced lesions give cats an appearance of old age because of the wrinkling of the skin of the face. See Table 1, page 128, for morphology.

Species and hosts:

Notoedres cati — Cat, fox (Figs. 219 to 223)
Notoedres cati var. *cuniculi* — Rabbit

(3) *Cnemidocoptic mites.* Scaly-leg and depluming scabies of birds are caused by mites of this genus. In the rather common disease, scaly-leg, the mites burrow under the scales of the legs and toes, causing dense crusts to form (Fig. 224). Scaly-leg mites are approximately 0.5 mm. in diameter. They are globular in shape. The legs of the adult female are very short; whereas the legs of the male are longer and are provided with suckers. See Table 1, page 128, and Fig. 202.

The depluming mite inhabits the skin at the bases of the feathers, especially around the head and neck. Infested birds pick out or scratch out the affected feathers because of the intense irritation. The morphology of depluming mites is much like that of scaly-leg mites, except that the size of the female is approximately 0.35 mm.

Species and hosts:

Cnemidocoptes mutans. Scaly-leg mite — Chicken, turkey, pheasant, caged birds (Figs. 225, 226)
Cnemidocoptes gallinae. Depluming mite — Chicken

Family 2. PSOROPTIDAE

(1) *Psoroptic mites.* The mites of this genus are the cause of sheep scab, cattle scab, and similar infestations on other hosts. They differ from the sarcoptic mites in morphology (Table 1, page 128) and in their manner of producing lesions. Psoroptic

mites do not burrow into the epidermis, but remain upon the surface or under scabs and scaly accumulations. In sheep, the thickly-wooled areas are attacked first. Itching is fairly pronounced and there is considerable loss of wool. In rabbits, a species of psoroptic mite infests the ear canals, resulting in severe otitis externa which is accompanied by thick scab formation. The psoroptic mites may be as large as 0.8 mm., hence they may be seen with or without the use of a hand lens. See Table 1, page 128, and Figs. 203 and 227 for morphology.

Species and hosts:

Psoroptes equi var. *equi* – Horse
Psoroptes equi var. *bovis* — Cattle (Fig. 228)
Psoroptes equi var. *ovis* — Sheep (Figs. 229 to 231)
Psoroptes equi var. *caprae* — Goat
Psoroptes equi var. *cuniculi* — Rabbit (Figs. 232, 233)

(2) *Chorioptic mites.* These were formerly known as symbiotic mites. They are the cause of so-called leg, foot, or tail mange. In heavy infestations the abdomen and other parts of the body are involved. Chorioptic mange is more common in the horse than in other domesticated animals. The lesions resemble those produced by psoroptic mites; in fact, the mites themselves are quite similar, except for the leg details (Table 1, page 128, and Figs. 204, 235) and for size. Chorioptic mites reach a maximum length of approximately 0.4 mm.

Species and hosts:

Chorioptes equi — Horse (Fig. 234)
Chorioptes bovis — Cattle (Figs. 236 to 239)
Chorioptes ovis — Sheep
Chorioptes caprae — Goat

(3) *Otodectic mites.* As their name implies, these mites invade the ear canals. They are parasites of dogs, cats, foxes, and other carnivora. Their presence is characterized by otitis externa, accompanied by bacterial decomposition of the secretions and of the exudate. Ear mites may be seen grossly or with the aid of an otoscope, their size being approximately 0.5 mm. in diameter.

For specific diagnostic features, see Table 1, page 128, and Fig. 205.

Species and hosts:

Otodectes cynotis. Ear mite — Dog, cat, fox, other carnivores (Figs. 240 to 243)

Family 3. EPIDERMOPTIDAE

This family contains two genera of uncommon skin mites infesting chickens, namely the genus Epidermoptes and the genus Rivoltasia, each including one species.

Epidermoptes bilobatus causes a rare form of avian scabies which is characterized by brownish-yellow, elevated scabs on the body and upper portions of the legs. The mites of both sexes have suckers on all of the leg-terminations. The length of the adult female is approximately 0.2 mm.

The other species of epidermoptic mite is *Rivoltasia bifurcata,* a feather-eating form, rarely reported from chickens. Apparently only slight damage is done to the infested feathers. These mites are approximately 0.25 mm. in length.

Species and hosts:

Epidermoptes bilobatus. Scaly skin mite — Chicken
Rivoltasia bifurcata. Feather-eating mite — Chicken

Family 4. CYTODITIDAE

This family of mites contains only one species, the air-sac mite of birds. Cytoditid mites belong to a small group of ectoparasites which have adapted their mode of living to the deeper tissues of the body. Therefore they are not, in a strict sense, skin parasites.

Cytodites nudus appears to be a fairly common inhabitant of the air-sacs, bronchi, lungs, and the bony cavities connected with the respiratory system. It is commonly called the air-sac mite. Hosts include chickens, turkeys, pigeons, and pheasants. Unless air-sac mites are abundant, they apparently do little harm; but in large numbers they may be associated with emaciation and anemia. Infected chickens have been known to show symptoms suggestive of avian tuberculosis. Close inspection of the air-sacs, soon after the host dies, is necessary in order to detect air-sac mites. They may be seen as minute transluscent dots, slowly moving about. These mites are less than 0.6 mm. in length. They resemble the sarcoptic mites.

Species and hosts:

Cytodites nudus. Air-sac mite — Chicken, turkey, pigeon, pheasant (Figs. 246, 247)

Family 5. LAMINOSIOPTIDAE

Laminosioptes cysticola is commonly called the subcutaneous mite or flesh mite of birds. Very little is known of its habits. Perhaps it is a skin parasite with a tendency to penetrate to the loose subcutaneous tissues, where it dies. The living mites are seldom observed, probably because they do not produce gross lesions until they die. Most frequently their presence is indicated by yellowish nodules several millimeters in diameter in the subcutis. These nodules appear to be caseo-calcareous enclosures around the mites, thus representing a defensive mechanism of the host. Subcutaneous mites are elongated, measuring approximately 0.25 mm. long by 0.1 mm. wide. A distinctive microscopic feature is the transverse constriction around the body posterior to the second pair of legs.

Species and hosts:

Laminosioptes cysticola. Subcutaneous mite — Chicken, turkey, goose, pigeon, pheasant

Family 6. DERMOGLYPHIDAE

These are uncommonly reported inhabitants of the feathers of birds, where they apparently feed, hence the name feather-eating mites.

(1) *The genus Falculifer.* One species, *Falculifer rostratus,* is a feather-damaging mite of pigeons. It is usually found between the barbs of the large wing feathers, causing the loss of barbules. Its length is approximately 0.5 mm.

Species and host:

Falculifer rostratus — Pigeon (Fig. 249)

(2) *The genus Freyana.* One species, *Freyana chaneyi,* has been reported from turkeys in Maryland, Texas, and Louisiana. It is said to congregate in the grooves under the shafts of the wing feathers. Little else is known about this mite.

Species and host:

Freyana chaneyi — Turkey

Family 7. ANALGESIDAE

The genus Megninia. This genus of analgesid feather mites is represented by three species in North American domesticated birds.

Megninia gallinulae has been reported only from Canada and then rarely. It is associated with loss of scales from the lower portions of the legs of chickens, and with a crusty dermatitis in the region of the head.

Megninia cubitalis is a similar mite which has been briefly mentioned as occurring on the feathers of chickens in southern United States. It is approximately 0.4 mm. in length.

Megninia columbae is approximately 0.3 mm. in length, and has been reported as occurring on the feathers of the neck and body of pigeons in South Carolina.

Species and hosts:

Megninia gallinulae — Chicken
Megninia cubitalis — Chicken, turkey
Megninia columbae — Pigeon

II. Suborder TROMBIDIFORMES

Family 1. DEMODICIDAE

These mites are the cause of demodectic, follicular, or red mange in a variety of hosts. The mites have a distinct appearance. The non-hairy body is elongated; the very short four pairs of legs are situated anteriorly; and the abdomen is transversely striated (Fig. 206). The adults are approximately 0.1 to 0.39 mm. in length. Demodectic mites live in the hair follicles and the sebaceous glands where they reproduce quite rapidly. Loss of hair is usually the first symptom of infestation, later to be followed by dermal hyperemia, and eventually by the formation of pustules. The latter are caused by secondary pyogenic bacterial infection.

Although demodectic mange is quite common in dogs, it may also occur in horses, cattle, sheep, goats, and swine. In these less common hosts the only observable lesions may be the formation of cutaneous nodules, varying in size up to 10 or 15 mm. in diameter. These nodules are filled by caseous pus containing an abundance of the mites.

Species and hosts:

Demodex equi — Horse
Demodex bovis — Cattle
Demodex canis var. *ovis* — Sheep
Demodex caprae — Goat
Demodex phylloides — Swine
Demodex canis — Dog (Figs. 244, 245)

Family 2. TROMBICULIDAE

This family includes the chigger mites, also called redbugs. Only the larval stage is parasitic; the adults and nymphs being free-living or predaceous on insects and other arthropods. Larval chiggers may infest the skin of many mammals, including man, and also the skin of many avian hosts. It is believed that their principle hosts are snakes, lizards, turtles, ground birds, and rabbits.

In attacking the host, chiggers insert the mouthparts (chelicerae) and inject a tissue-liquefying saliva. Within a few hours intense pruritus with swelling occurs. The pruritus lasts for days to weeks. Chiggers do not bodily enter the skin while feeding on liquefied tissues. Usually after several hours' attachment they release their hold and drop to the ground for further development. Larval chiggers are difficult to detect on animals. They vary in color from yellowish to red and their length is about 0.45 mm.

Species and hosts:

Eutrombicula (= *Trombicula*) *alfreddugési*. North American chigger — Various mammals and birds (Fig. 248)

Neoschöngastia americana. Chicken chigger — Chicken, other birds, rabbits, lizards, snakes. Found in southern United States.

Family 3. MYOBIIDAE

(1) *Syringophilus bipectinatus,* a quill mite, is an inhabitant of the quills of domesticated and wild birds. Its presence is indicated by a powdery accumulation inside the quills of the larger feathers, causing their partial to complete loss. The adult female measures about 0.9 mm. in length by about 0.15 mm. in width. It is seldom reported.

(2) *Psorergates ovis,* a so-called itch mite of sheep, was first reported by Carter (1941) in Australia. Its first occurrence in North America was noted by Bell *et al.* in Ohio in 1952. Davis (1954) has also studied the sheep itch mite.

Infested sheep rub, scratch, or bite at the wool because of a mild chronic dermatitis. Tags of wool hang from the fleece or drop off.

Psorergates mites have legs more or less equidistant apart, whereas the legs of the common mange and scab mites are in groups of two. The adult itch mite of sheep may be as large as 0.189 by 0.162 mm.

Species and host:

Syringophilus bipectinatus. Quill mite — Chicken, turkey, pheasant, other birds

Psorergates ovis. Sheep itch mite — Sheep

Family 4. CHEYLETIDAE

Mites of this family are elongated and possess pincer-like feather-clasping organs (palpi) on each side of the mouthparts. Most of the cheyletid mites are free-living predators of insects or of other mites. One species, *Cheyletiella parasitivorax,* has been reported from the skin of cats and rabbits of North America in recent years (Cooper, 1946; Roth, 1947).

This mite may be found in large numbers in the fur. In North America no gross lesions have been attributed to its presence. Cheyletid dermatitis of cats and humans has been reported in Europe. Probably this mite preys upon parasitic mange mites. It has also been found attached to fleas, possibly as a means of transportation. The adults are about 0.45 mm. long.

Species and hosts:

Cheyletiella parasitivorax — Cat, rabbit

Family 5. SPELEOGNATHIDAE

A speleognathid mite, *Speleognathus striatus,* was reported in North America from the nasal cavity of the domestic pigeon by Crossley (1952). Its pathogenicity is unknown. Probably it is transmitted through contaminated drinking utensils. The length is about 0.5 mm.

Species and host:

Speleognathus striatus. A nasal mite — Pigeon

III. Suborder MESOSTIGMATA

Family 1. DERMANYSSIDAE

Two genera of this family, Dermanyssus and Bdellonyssus, contain parasites of domesticated birds.

(1) *The genus Dermanyssus.* One important species, *Dermanyssus gallinae,* is the common chicken mite (red mite, roost mite). Its hosts include chickens, turkeys, pigeons, English sparrows, and other birds. Man and other mammals may be attacked if the mites are abundant. This mite has needle-like mouthparts for sucking blood. Red mites breed in the hosts' surroundings, attacking mostly at night or when the birds are nesting. Adult females, engorged with blood, may reach a length of 1 mm.

Species and hosts:

Dermanyssus gallinae. Common red mite — Chicken, turkey, pigeon, other birds, occasionally mammals (Fig. 250)

(2) *The genus Bdellonyssus* (= *Liponyssus*). Three species of feather mites have been reported from North America. Although resembling mites of the preceding genus, they differ mainly in that they are found on their bird hosts both day and night, where they suck blood.

The most common feather mite is *Bdellonyssus sylviarum,* or Northern feather mite. A second species, *Bdellonyssus canadensis,* was reported from Canada by Hearle (1938). A third species, *Bdellonyssus bursa,* the tropical feather mite, occurs in the South Atlantic and South Central states. Many birds, in addition to chickens are reported to harbor these mites. Adult feather mites are about 0.7 mm. in length.

Species and hosts:

Bdellonyssus sylviarum. Northern feather mite — Chicken and many other bird species (Fig. 251)

Bdellonyssus canadensis. Canadian feather mite — Chicken and other bird species

Bdellonyssus bursa. Tropical feather mite — Chicken and other bird species

Family 2. RAILLIETIDAE

One species of mite belonging to this family has been rarely reported from cattle in North America. Probably it is more common than the records show. Leidy in 1872 found *Raillietia auris* in the external ear canal of cattle near Philadelphia. It was not until 1950 that it was again reported, this time by Olsen and Bracken in Colorado. Benbrook (unpublished data), in 1925, identified this mite from the ear canals of a steer that had been shipped into Iowa from Minnesota. This steer showed incoordination and apathy. At necropsy, the mites were seen moving rapidly over and near the tympanic membrane. No other evidence was found to account for the symptoms. The adults are approximately 1.5 mm. in length.

Species and host:

Raillietia auris. Ear mite — Cattle (Fig. 252)

Family 3. HALARACHNIDAE

The mites of this family occur in the respiratory passages of marine mammals (seals, walruses) and land mammals (carnivores, monkeys, rodents).

One species, *Pneumonyssus caninum,* is of interest to the veterinarian. This mite occurs quite commonly in the frontal sinuses of dogs. Chandler and Ruhe (1940) first described it as a new species. Later references are those of Martin and Deubler (1943), Douglas (1951), Koutz *et al.* (1953), Olds (1953), and Furman (1954).

As yet its significance as a pathogen is not clear. Catarrhal or purulent sinusitis is often observed in the affected dogs. No nymphal stage is known. The mature mites are white, and 1 mm. long.

Species and host:

Pneumonyssus caninum. Frontal sinus mite – dog (Fig. 253)

Family 4. RHINONYSSIDAE

Rhinonyssid mites are parasitic in the nasal passages of various birds. Two species, *Neonyssus columbae* and *Neonyssus melloi,* have been reported in pigeons from Texas by Crossley (1950 and 1952). No further information is available. These mites are viviparous, producing larvae in which the nymphs are already developed. The adult length is about 0.7 mm.

Species and host:

Neonyssus columbae. Nasal mite – Pigeon
Neonyssus melloi. Nasal mite – Pigeon

Apparatus and Technique for the Examination of the Skin To Detect Parasitic Mites

Some species of mites that live on the skin, also those that inhabit the internal organs, can usually be seen with the unaided eye. A hand lens, of x 3 or greater magnification, is a useful agent for detection when used in a bright light. Any mites seen may be placed in a drop of water on a microslide. Then a coverglass is applied and the preparation is examined under low power (x 100) and high power (x 400) of the microscope. The substage condenser and the diaphragm are adjusted so as to provide a relatively low degree of light in order to reveal details of structure.

For the detection and identification of the various species of mange and scab mites, it is advisable to make scrapings of the skin, using the following apparatus and technique:

APPARATUS FOR SKIN SCRAPINGS (FIG. 191)

1. *The microscope.* Magnifications of approximately x 100 and x 410 are most suitable for the detection of skin mites. Therefore, the optical equipment should include an 8X or 10X Huyghenian ocular, 16 mm. and 4 mm. achromatic objectives, and a substage condenser of 1.25 numerical aperture. A mechanical stage and a binocular body tube with matched

FIG. 191—Apparatus for microscopic examination of skin preparations for animal parasites:

1. Microscope
2. Xylene
3. Lens paper
4. Microscope lamp
5. Coverglass forceps
6. Scalpel
7. Coverglasses
8. Microslides
9. Black paper
10. Mineral oil dispensers
11. Ear swab
12. Hand magnifier
13. Jar for waste
14. Towel

oculars are not essential, but they will save the examiner's time and help to reduce eyestrain. The addition of an oil immersion objective will equip the microscope for all the important clinical procedures that require microscopy.

2. *Xylene.* This is the only safe lens-cleaning solvent, except water. It should be dispensed from a dropper-bottle.
3. *Lens paper.* This is essential for keeping optical lenses clean. Squares of about 8 cm. (3 in.) may be stored in a covered container. They should be used once, then discarded.
4. *Microscope lamp.* Daylight should not be relied upon. There are many suitable types of microscope lamps. A simple type to be recommended consists of a metal shade enclosing a 60 watt, inside-frosted, blue bulb.
5. *Coverglass forceps.* These should always be used when handling micro coverglasses.

6. *Scalpel.* A detachable-blade surgical scalpel is preferred for scraping the skin. The blade should be convexly curved.
7. *Coverglasses.* Any 18 mm. or 22 mm. (3/4 or 7/8 in.) square, glass or plastic coverglass is suitable. The plastic covers are more economical and they require no cleaning before they are used, after which they are discarded. Coverglasses should be stored in a covered container, such as a small glass dish.
8. *Microslides.* These are the standard 75 x 25 mm. (3 x 1 in.) glass slides. They should be washed and dried before using, and they may be used repeatedly.
9. *Black paper.* A sheet of dull-surfaced black paper is used as a background in preparing the specimens on the microslides.
10. *Mineral oil and dispensers.* Any light-bodied mineral oil may be used to prepare the skin scraping. It may be dispensed from a dropper-bottle or from a small lubricating oilcan.
11. *Ear swabs.* Wooden applicator sticks 15 cm. (6 in.) in length are tipped with absorbent cotton for the removal of specimens from ear canals.
12. *Hand magnifier.* This should provide a magnification of **x** 3, or greater, for the examination of skin parasites, ear canal surfaces, or ear swabs.
13. *Jar for waste.* Skin mites may live for hours in mineral oil or in water. Discarded slides and swabs may be placed in a jar containing a disinfectant, such as 3 per cent aqueous saponified cresol solution.
14. *Towels.* Soft linen or cotton towels are used for cleaning the hands and equipment.

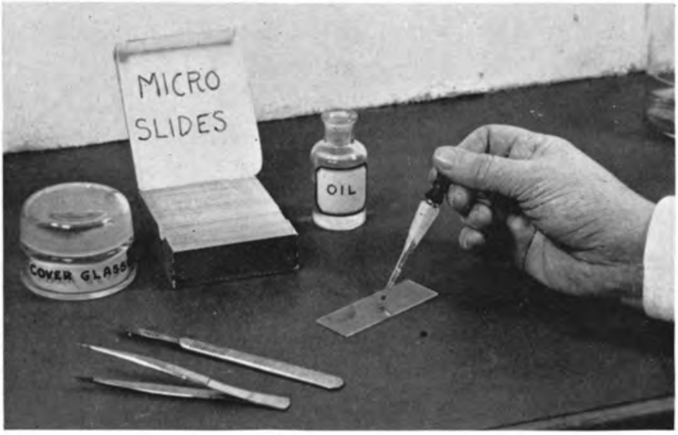

FIG. 192—Placing a drop of mineral oil on a microslide.

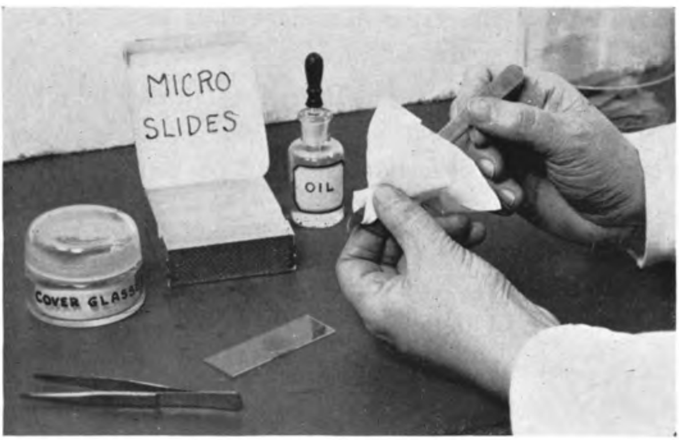

FIG. 193—Cleaning the scalpel blade.

FIG. 194—Dipping the cleaned scalpel blade into the drop of mineral oil before scraping the skin.

FIG. 195—Scraping a fold of a suspected facial lesion with the oiled scalpel blade.

FIG. 196—Scraping a fold of a suspected lesion on the leg.

FIG. 197—Transferring the scraping from the scalpel blade to the drop of oil on the microslide.

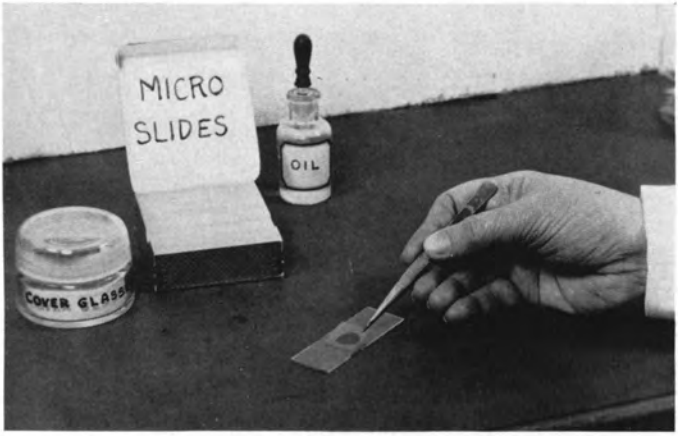

FIG. 198—Applying the coverglass, using forceps.

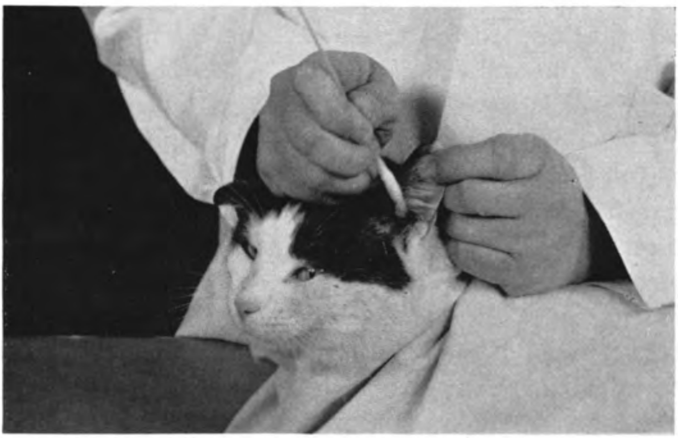

FIG. 199—Removing ear mites on a dry cotton swab. The patient is under restraint in a canvas roll.

FIG. 200—A black paper background and a hand magnifier are used in examining the cotton swab for ear mites.

TECHNIQUE FOR SKIN SCRAPINGS

1. Place a drop of mineral oil on a microslide (Fig. 192).
2. Clean the scalpel blade by wiping it with paper (Fig. 193).
3. Dip the clean scalpel blade into the drop of oil on the microslide (Fig. 194).
4. Pick up a fold of the patient's skin at the edge of the suspected area, pinching it firmly between the thumb and forefinger. With the oily scalpel, scrape the crest of the fold several times in the same direction. Scrapings will adhere to the blade. Stop scraping when a slight amount of blood appears (Figs. 195 and 196).
5. Transfer the scraping from the scalpel blade into the drop of oil on the microslide, using a slight rotary motion (Fig. 197).
6. Apply a coverglass to the scraping on the microslide by gently lowering it by means of a coverglass forceps. Additional oil may be added at the coverglass edge in order to fill the space beneath it. Do not press on the coverglass (Fig. 198).
7. Examine the preparation under low power (x 100) in a methodical manner so that all portions of the coverglass area are seen (Fig. 14). For best results, manipulate the substage condenser and diaphragm of the microscope so as to provide a relatively low degree of light, evenly distributed.

Oily preparations of mites may be kept for days as demonstration specimens. The mites show motion for many hours.

8. For the detection of ear mites in the dog, cat, fox, and rabbit, the patient may be restrained in a canvas sheet (Fig. 199). A cotton swab is introduced into the external auditory canal and gently rotated. The swab is then placed on a piece of black paper and examined by means of a hand lens (Fig. 200). Living and dead ear mites may be seen. If necessary, individual ear mites may be transferred on the tip of the scalpel blade from the cotton swab to a drop of oil on a microslide for microscopic examination. For best results a coverglass should be applied.

An electrically illuminated otoscope may be introduced directly into the ear canal for the detection of ear mites, thus making microscopic examination unnecessary.

The more rapidly-moving, larger skin mites may be captured by touching them with an oily cotton swab. This slows them down so that they may then be transferred to a drop of oil on a microslide for microscopic examination.

References for Section One will be found starting on page 169.

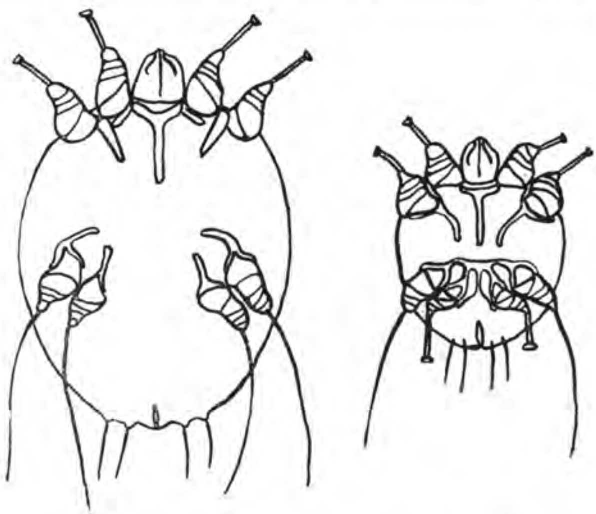

FIG. 201—Female and male mites of the genus Sarcoptes, drawn to show the diagnostic features listed in Table 1.

TABLE 1
MICROSCOPIC CHARACTERISTIC OF THE SARCOPTIFORM MANGE AND SCAB MITES

Group	Leg Characteristics		Anus
	Egg-laying Female	Male	
SARCOPTIC	Suckers on a long *unjointed* pedicle on pairs 1 and 2, Fig. 201	Suckers on a long *unjointed* pedicle on pairs 1, 2, and 4, Fig. 201	Terminal
NOTOEDRIC	As above	As above	Dorsal
CNEMIDO-COPTIC	No suckers, Fig. 202	Suckers on an *unjointed* pedicle on pairs 1, 2, 3 and 4, Fig. 202	Terminal
PSOROPTIC	Suckers on a long *jointed* pedicle on pairs 1, 2, and 4, Fig. 203	Suckers on a long *jointed* pedicle on pairs 1, 2, and 3, Fig. 203	Terminal
CHORIOPTIC	Suckers on a short *unjointed* pedicle on pairs 1, 2, and 4, Fig. 204	Suckers on a short *unjointed* pedicle on pairs 1, 2, 3, and 4. Pair 4 rudimentary. Fig. 204	Terminal
OTODECTIC	Suckers on a short *unjointed* pedicle on pairs 1 and 2. Pair 4 rudimentary. Fig. 205	Suckers on a short *unjointed* pedicle on pairs 1, 2, 3, and 4, Fig. 205	Terminal

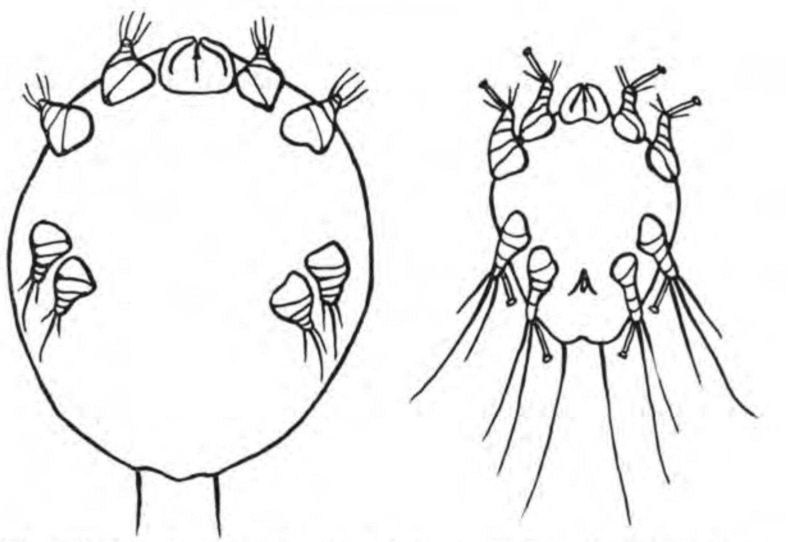

FIG. 202—Female and male mites of the genus Cnemidocoptes, drawn to show the diagnostic features listed in Table 1.

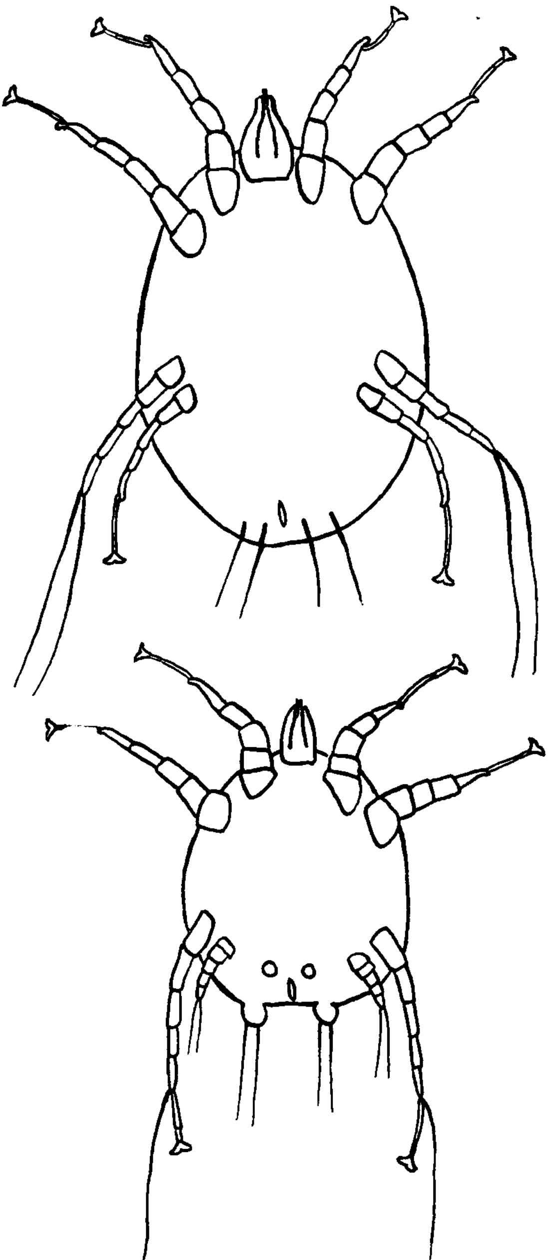

FIG. 203—Female and male mites of the genus Psoroptes, drawn to show the diagnostic features listed in Table 1.

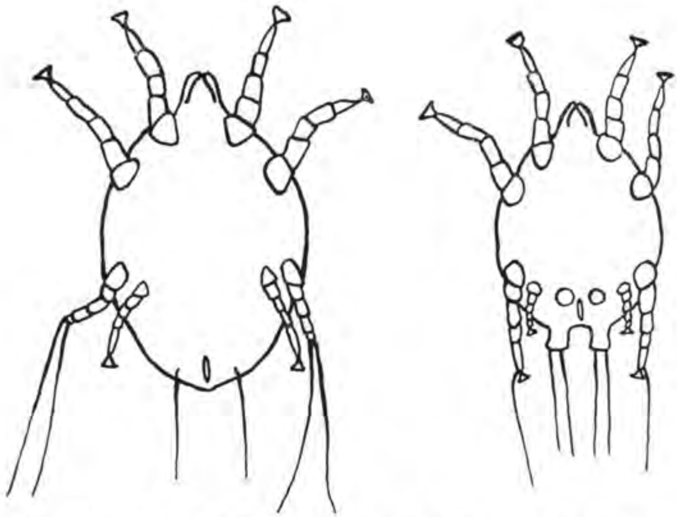

FIG. 204—Female and male mites of the genus Chorioptes, drawn to show the diagnostic features listed in Table 1.

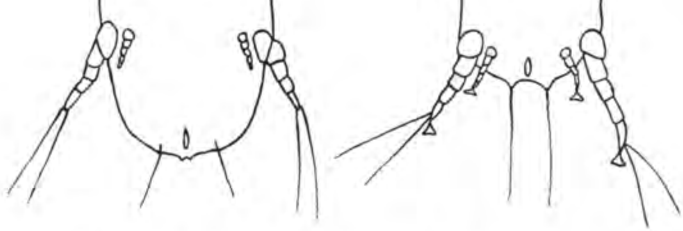

FIG. 205—Female and male mites of the genus Otodectes, drawn to show the diagnostic features listed in Table 1.

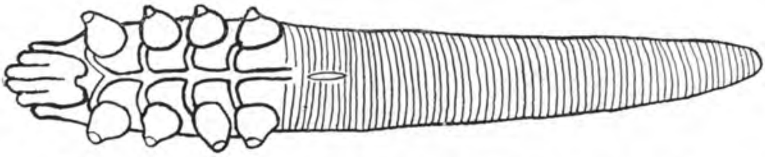

FIG. 206—Female mite of the genus Demodex, drawn to show the diagnostic features.

CATTLE

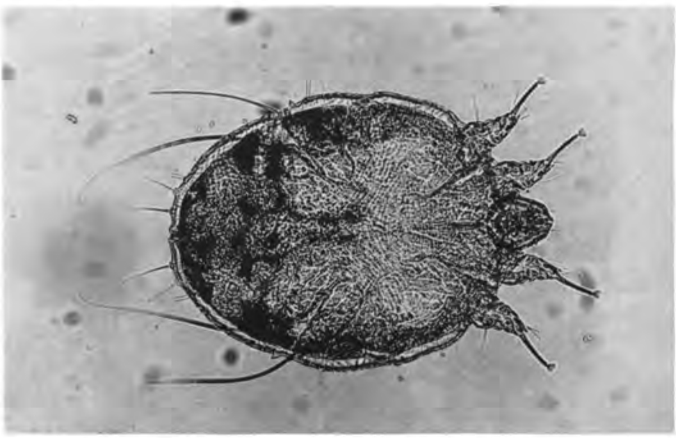

FIG. 207—Adult female **Sarcoptes scabiei** var. **bovis,** the sarcoptic mange mite of cattle. x 130.

SWINE

FIG. 208—Sarcoptic mange lesion on the hind quarter of a pig.

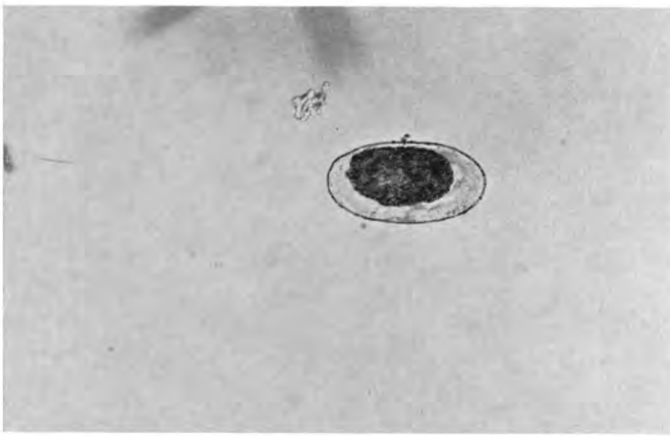

FIG. 209—Ovum of **Sarcoptes scabiei** var. **suis,** the sarcoptic mange mite of swine. x 100.

SWINE

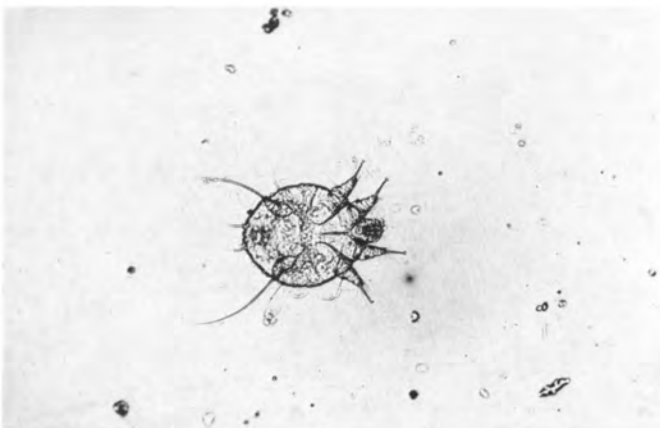

FIG. 210—Larval **Sarcoptes scabiei** var. **suis,** the sarcoptic mange mite of swine. x 100.

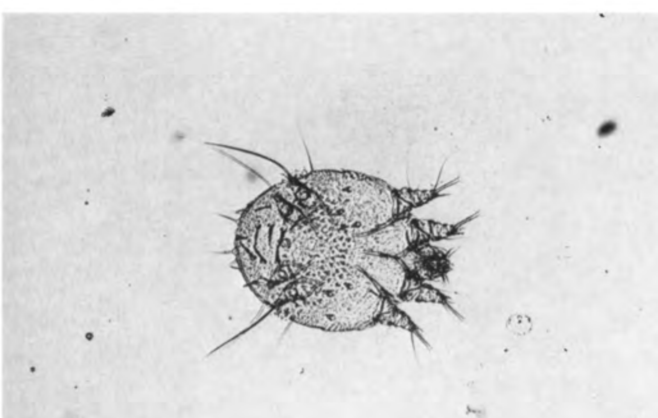

FIG. 211—Nymph of **Sarcoptes scabiei** var. **suis,** the sarcoptic mange mite of swine. x 100.

SWINE

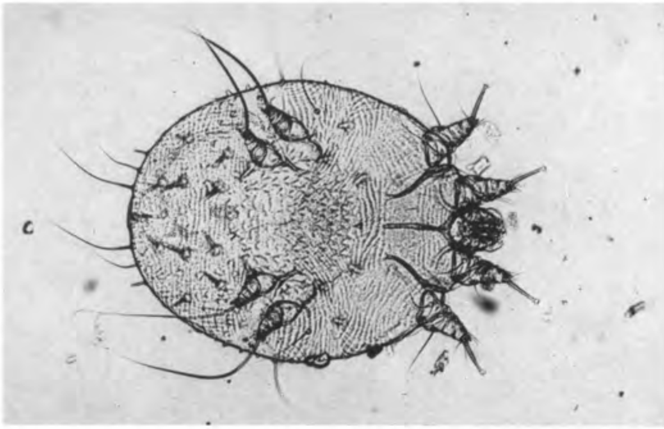

FIG. 212—Adult female **Sarcoptes scabiei** var. **suis,** the sarcoptic mange mite of swine. x 100.

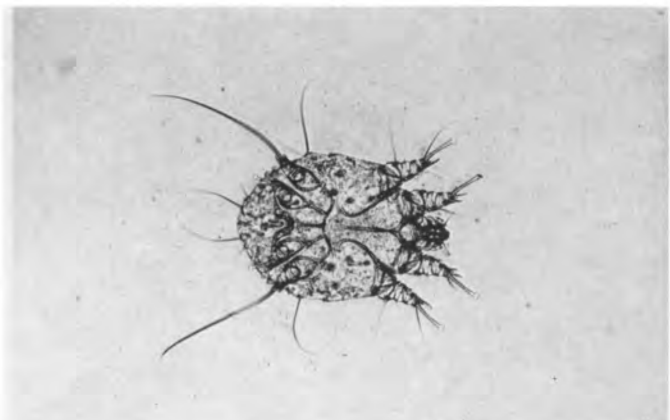

FIG. 213—Adult male **Sarcoptes scabiei** var. **suis,** the sarcoptic mange mite of swine. x 100.

DOG

FIG. 214—Ova of **Sarcoptes scabiei** var. **canis,** the sarcoptic mange mite of dogs. x 100.

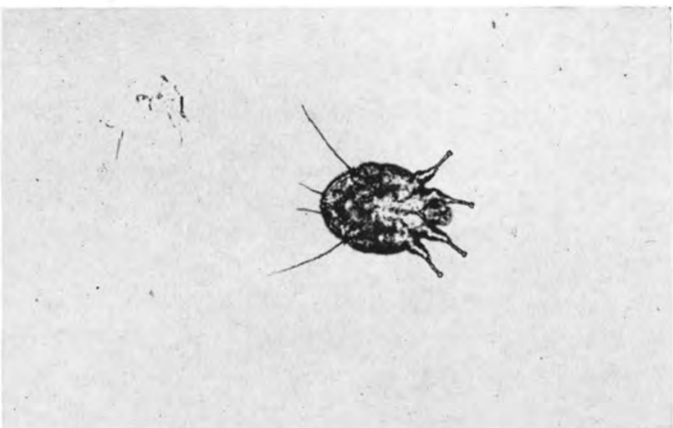

FIG. 215—Larval **Sarcoptes scabiei** var. **canis,** the sarcoptic mange mite of dogs. x 100.

DOG

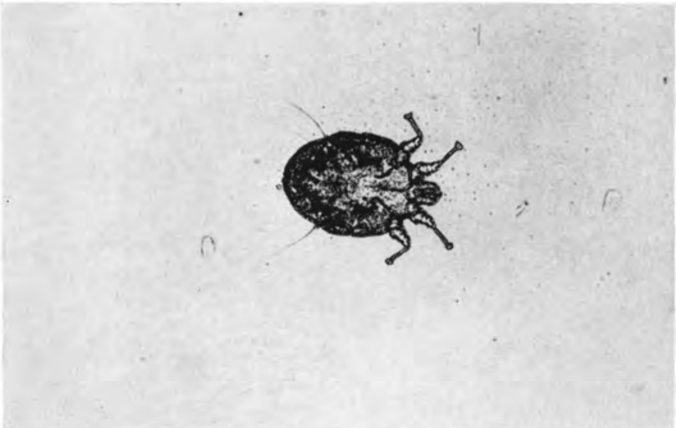

FIG. 216—Nymph of **Sarcoptes scabiei** var. **canis,** the sarcoptic mange mite of dogs. x 100.

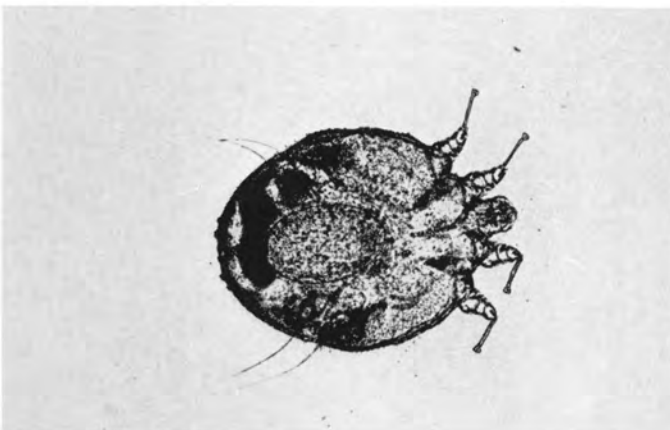

FIG. 217—Adult female **Sarcoptes scabiei** var. **canis,** the sarcoptic mange mite of dogs. x 100.

DOG

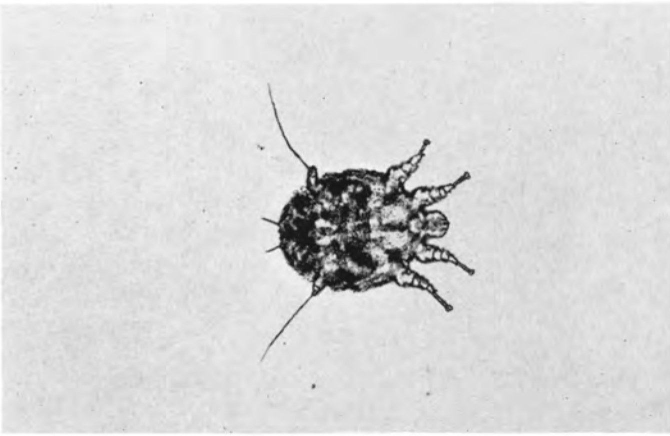

FIG. 218—Adult male **Sarcoptes scabiei** var. **canis,** the sarcoptic mange mite of dogs. x 100.

CAT, FOX, RABBIT

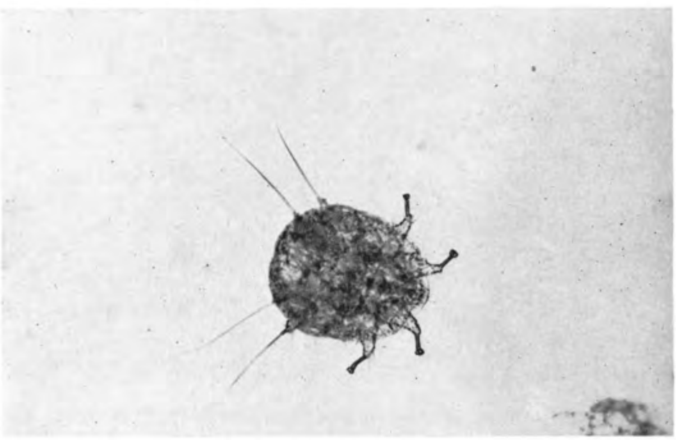

FIG. 219—Adult female **Notoedres cati,** the notoedric mange mite of cats, foxes, and rabbits. x 100.

CAT, FOX, RABBIT

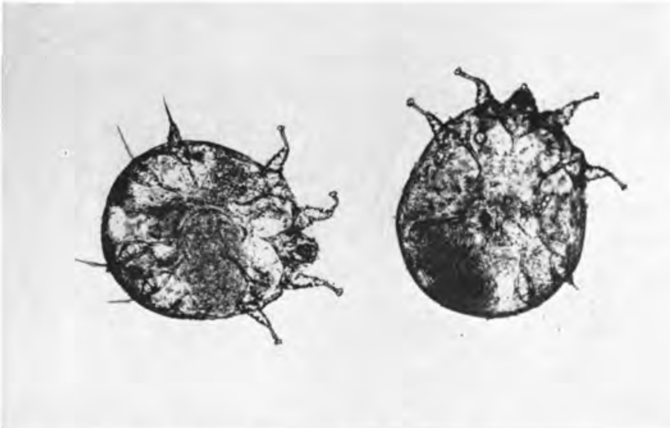

FIG. 220—Adult female **Notoedres cati,** the notoedric mange mite of cats, foxes, and rabbits. x 110.

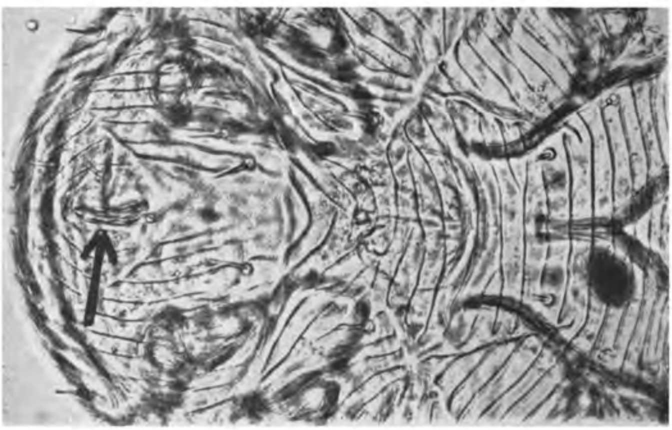

FIG. 221—Posterior dorsal abdomen of **Notoedres cati,** the notoedric mange mite of cats, foxes, and rabbits. The arrow shows the slitlike anus, located dorsally rather than terminally as in the genus Sarcoptes. x 410.

FOX

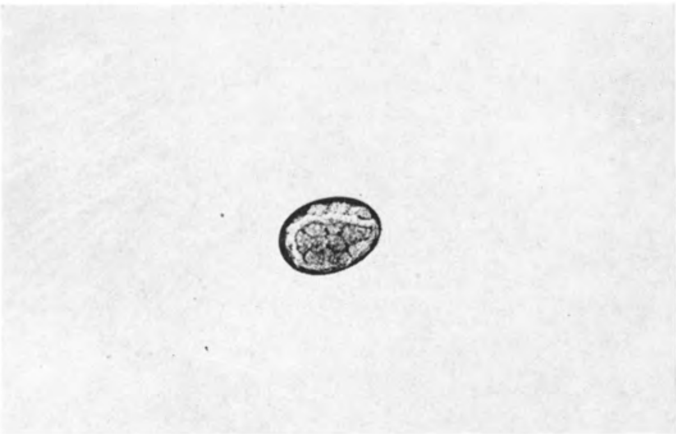

FIG. 222—Ovum of **Notoedres** sp., a nctoedric mange mite of foxes. x 110.

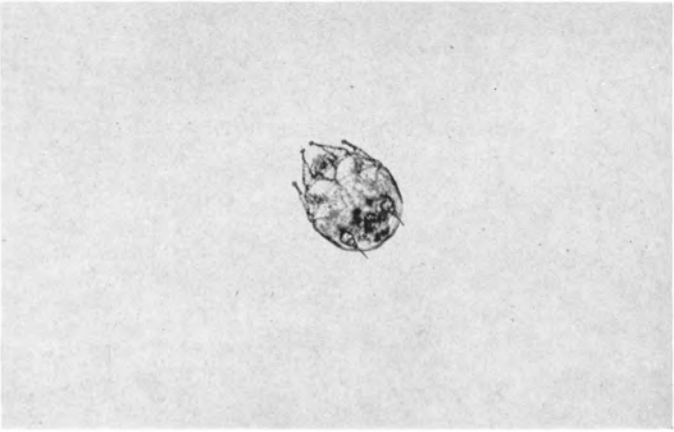

FIG. 223—Larva of **Notoedres** sp., a notoedric mange mite of foxes. x 110.

CHICKEN

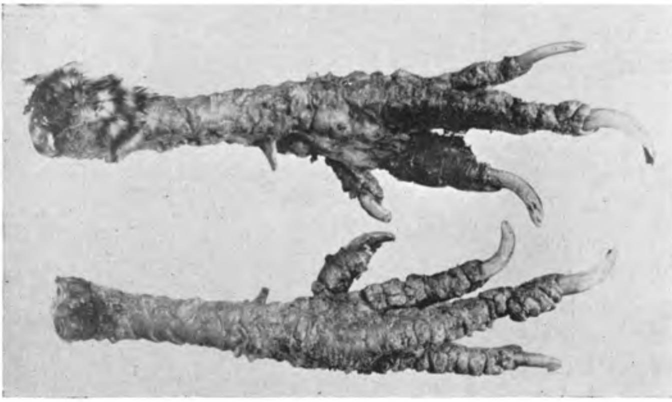

FIG. 224—Lesions of scaly-leg of poultry, caused by **Cnemidocoptes mutans.**

CHICKEN

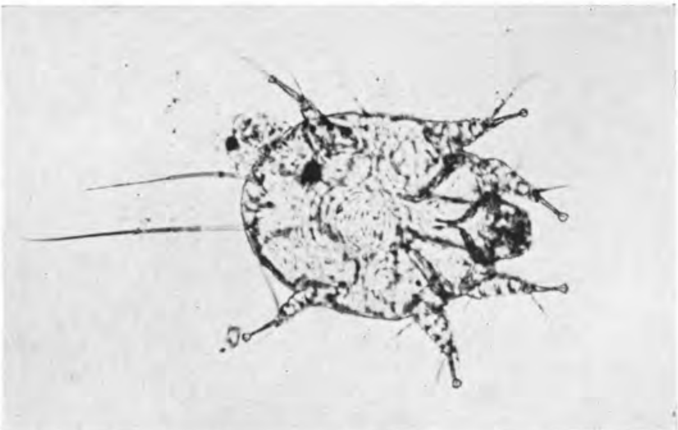

FIG. 225—Larva of **Cnemidocoptes mutans,** the scaly-leg mite of poultry. x 200.

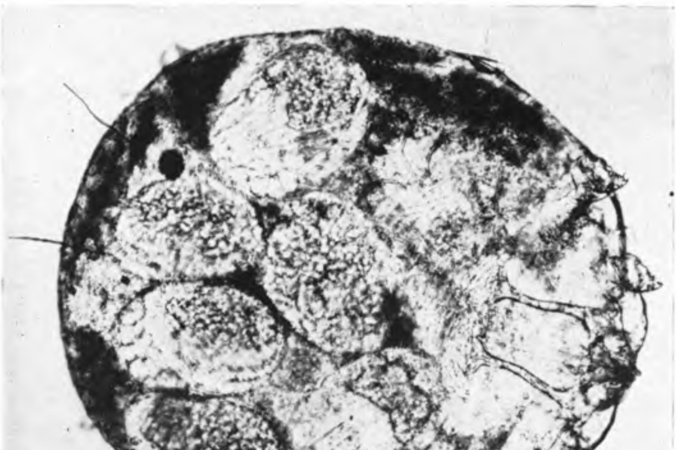

FIG. 226—Adult female **Cnemidocoptes mutans,** the scaly-leg mite of poultry. x 145.

CATTLE

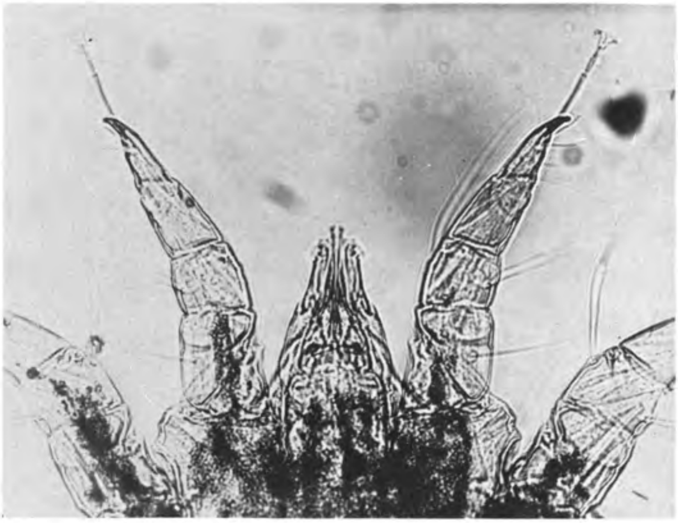

FIG. 227—Leg detail of **Psoroptes equi** var. **bovis.** The suckers are on long jointed pedicles. x 188.

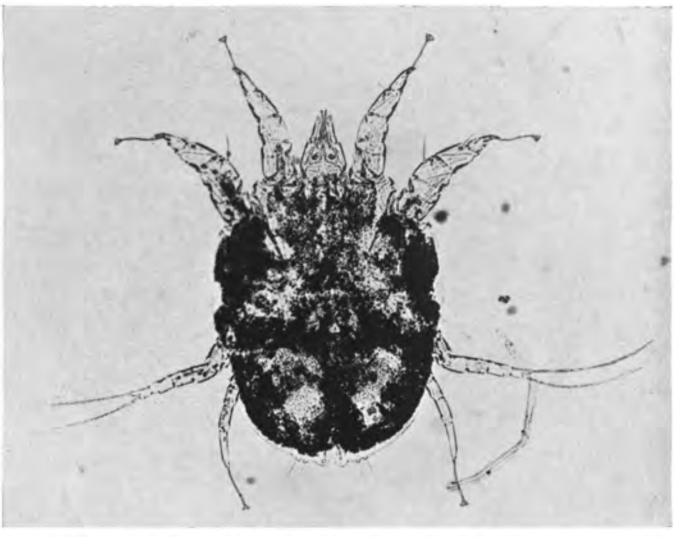

FIG. 228—Adult female **Psoroptes equi** var. **bovis,** the psoroptic or scab mite of cattle. x 80.

SHEEP

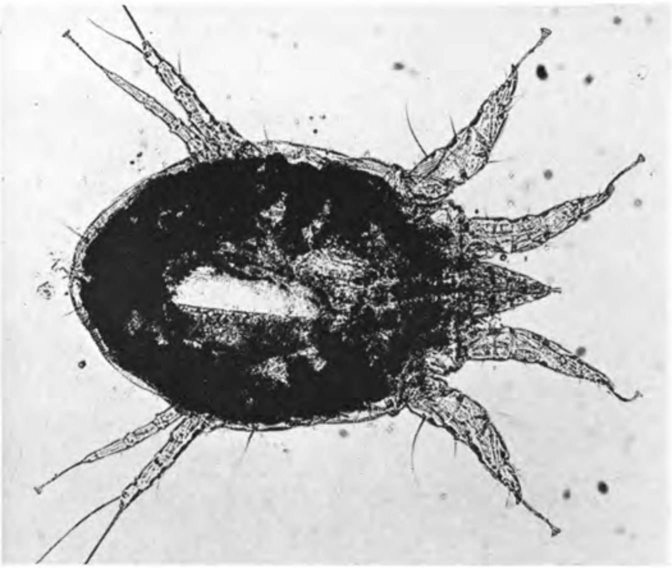

FIG. 229—Ovigerous female **Psoroptes equi** var. **ovis,** the scab mite of sheep. x 90.

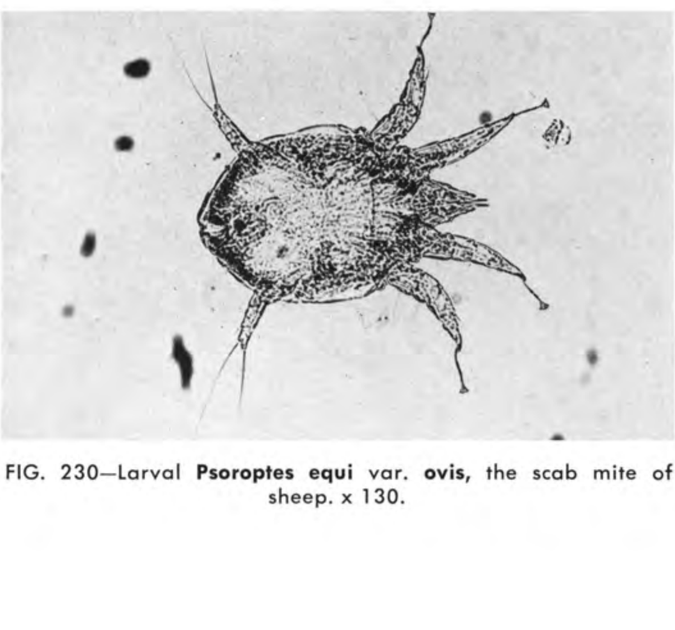

FIG. 230—Larval **Psoroptes equi** var. **ovis,** the scab mite of sheep. x 130.

SHEEP

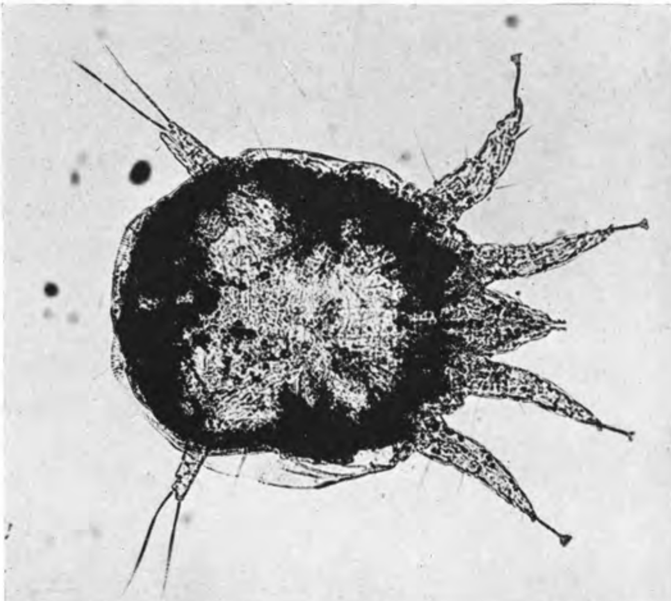

FIG. 231—Pubescent female **Psoroptes equi** var. **ovis,** the scab mite of sheep. The posterior pairs of legs are shortened until after copulation. x 120.

RABBIT

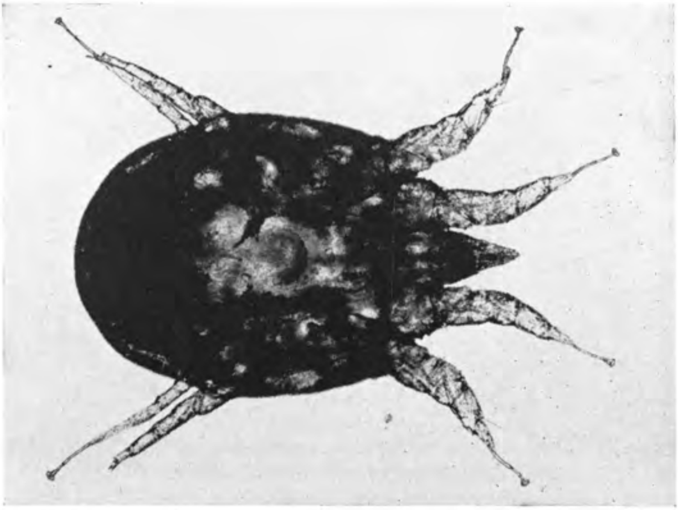

FIG. 232—Adult female **Psoroptes equi** var. **cuniculi,** an ear scab mite of rabbits. x 75.

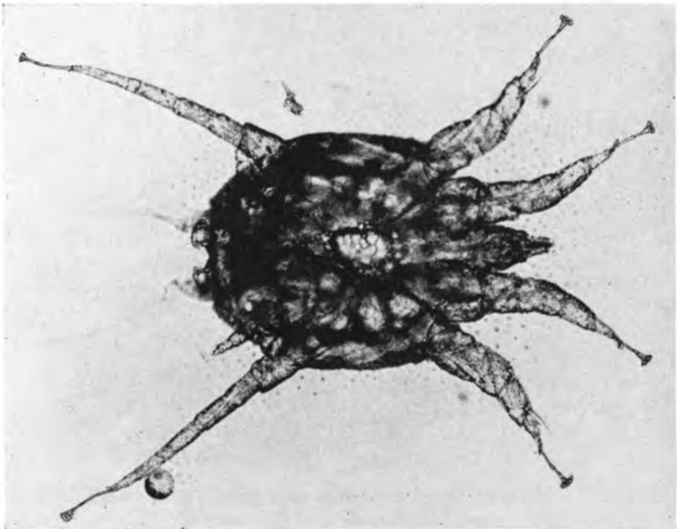

FIG. 233—Adult male **Psoroptes equi** var. **cuniculi,** an ear scab mite of rabbits. x 75.

HORSE

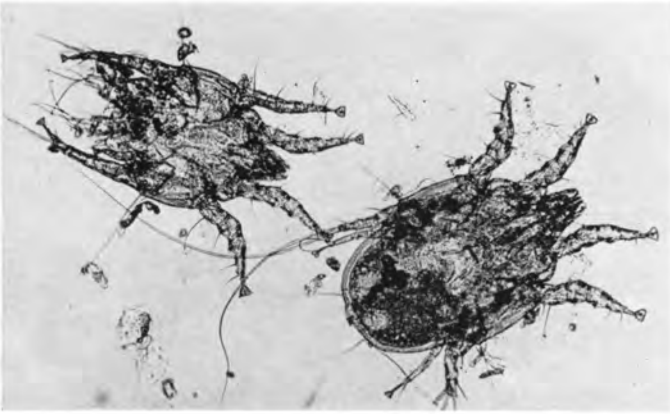

FIG. 234—Adult male **(left)** and female **(right)** **Chorioptes equi,** the chorioptic mange mite of horses. x 90.

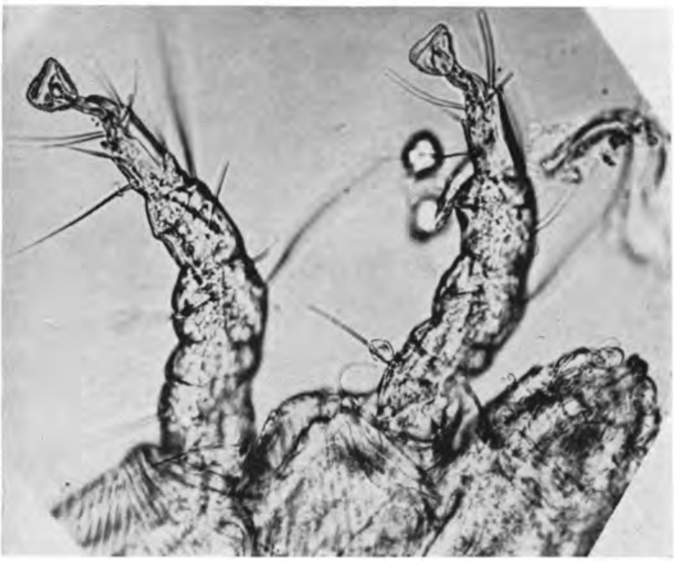

FIG. 235—Leg detail of **Chorioptes equi.** The suckers are on short, unjointed pedicles. x 350.

CATTLE

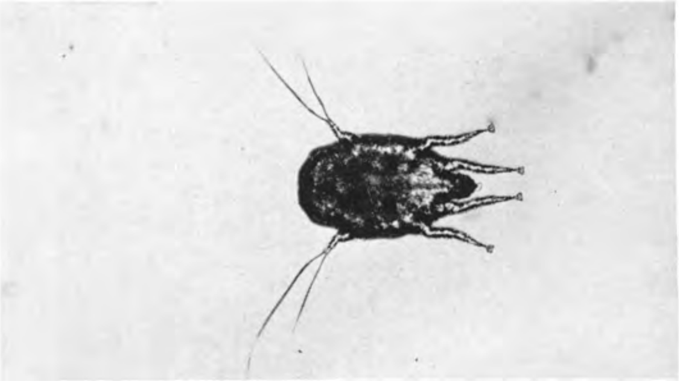

FIG. 236—Larva of **Chorioptes bovis,** the chorioptic mange mite of cattle. Note that there are only three pairs of legs in the larval stage of mites. x 100.

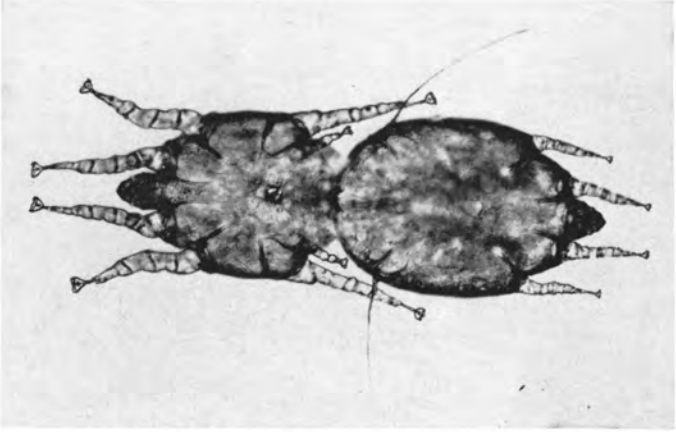

FIG. 237—**Chorioptes bovis,** the chorioptic mange mite of cattle, in copulation. x 100.

CATTLE

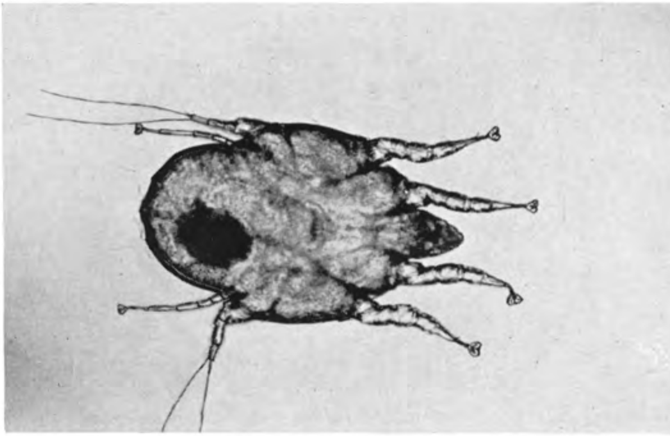

FIG. 238—Adult female **Chorioptes bovis,** the chorioptic mange mite of cattle. x 100.

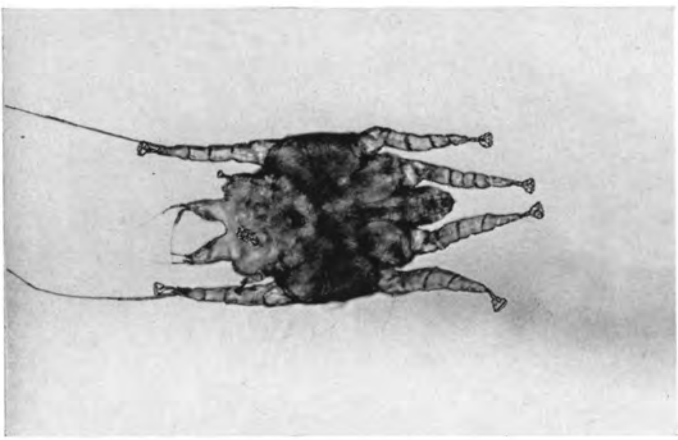

FIG. 239—Adult male **Chorioptes bovis,** the chorioptic mange mite of cattle. x 100.

DOG, FOX, CAT

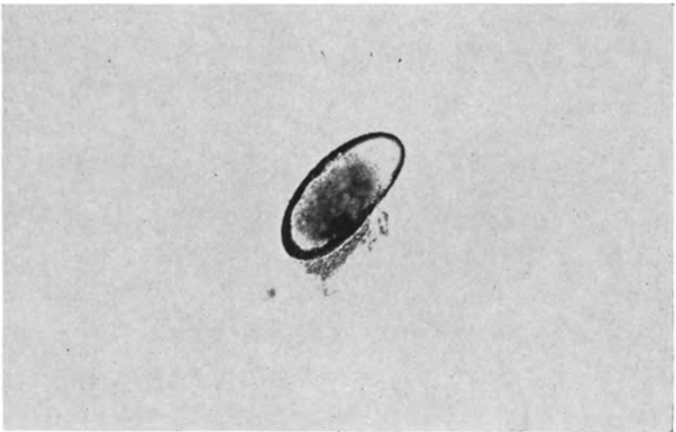

FIG. 240—Ovum of **Otodectes cynotis,** the ear mange mite of dogs, foxes, and cats. x 100.

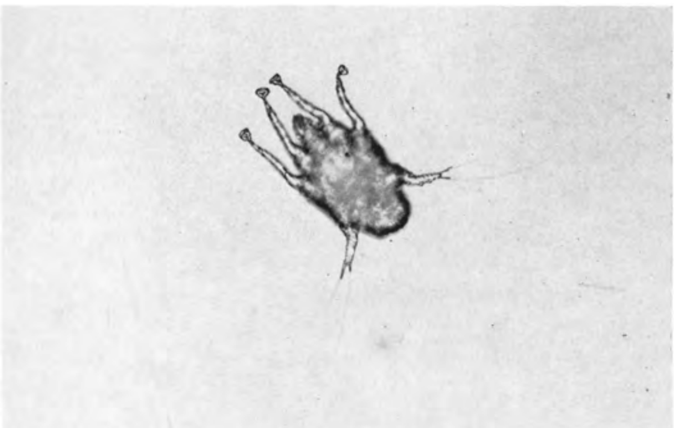

FIG. 241—Larva of **Otodectes cynotis,** the ear mange mite of dogs, foxes, and cats. x 100.

DOG, FOX, CAT

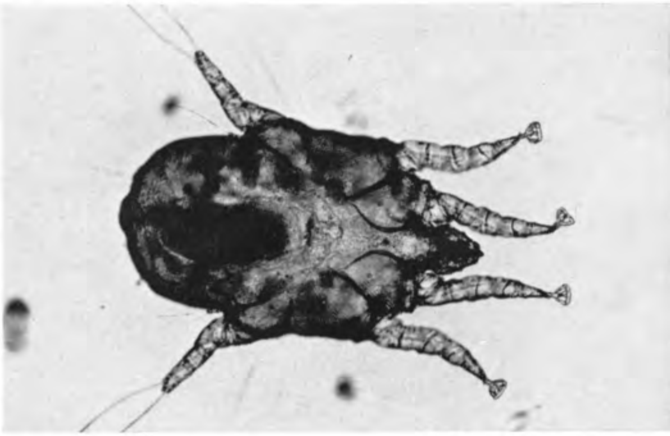

FIG. 242—Adult female **Otodectes cynotis,** the ear mange mite of dogs, foxes, and cats. x 100.

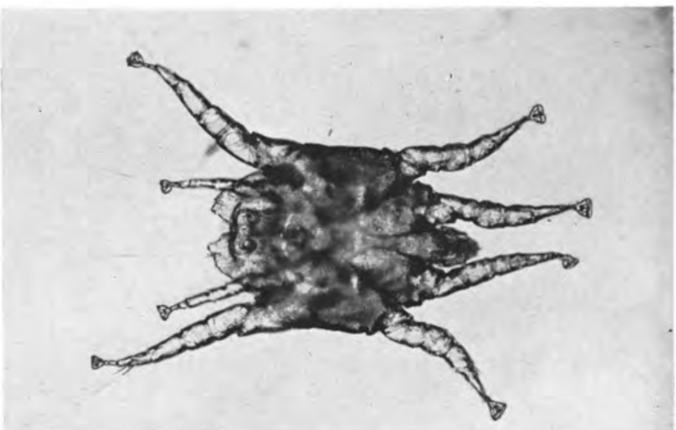

FIG. 243—Adult male **Otodectes cynotis,** the ear mange mite of dogs, foxes, and cats. x 100.

DOG

FIG. 244—Adults and an ovum **(right)** of **Demodex canis,** the demodectic mange mite of dogs. x 100.

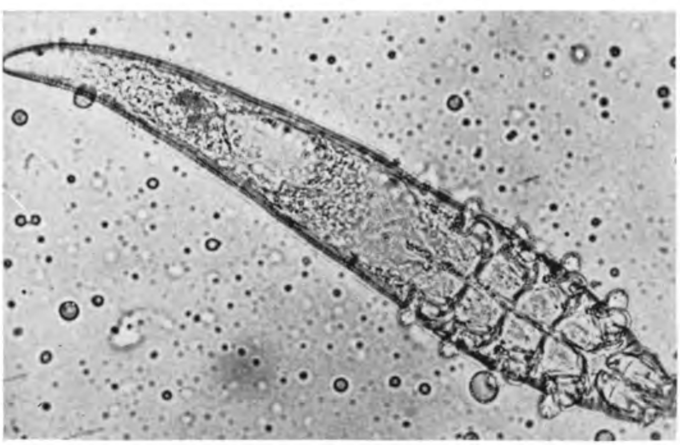

FIG. 245—Adult female **Demodex canis,** the demodectic mange mite of dogs. x 410.

POULTRY

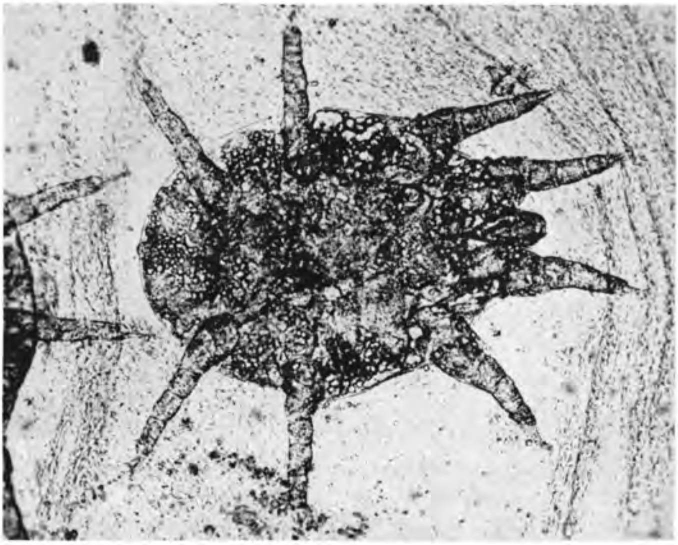

FIG. 246—Adult female **Cytodites nudus,** the air-sac mite of poultry. A portion of an air-sac appears in the background. x 100.

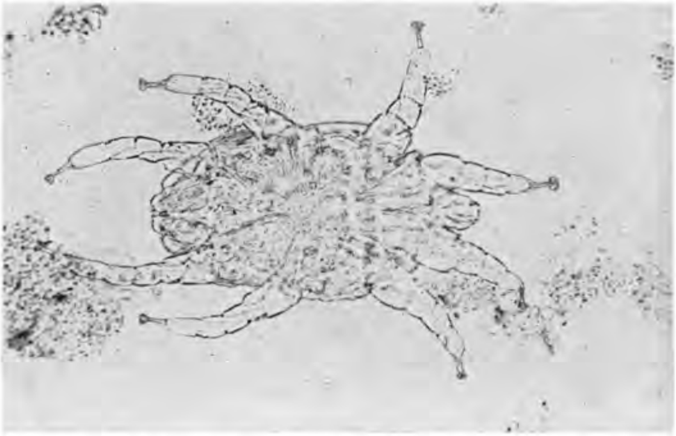

FIG. 247—Adult male **Cytodites nudus,** the air-sac mite of poultry. x 100.

MAMMALS, POULTRY

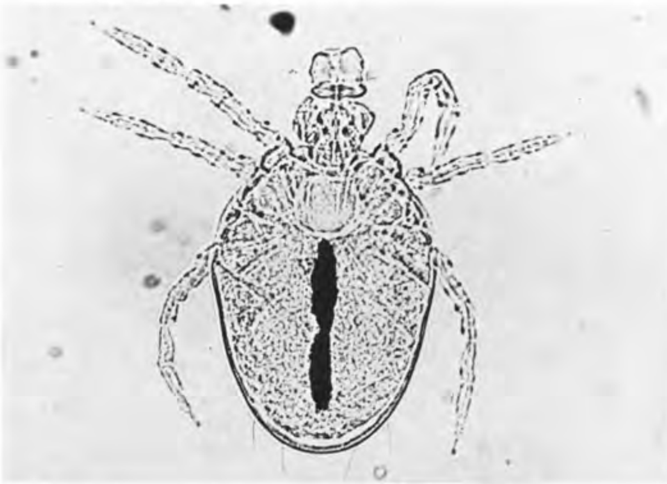

FIG. 248—Larva of **Eutrombicula alfreddugési,** the chigger mite of mammals and poultry. x 130.

PIGEON

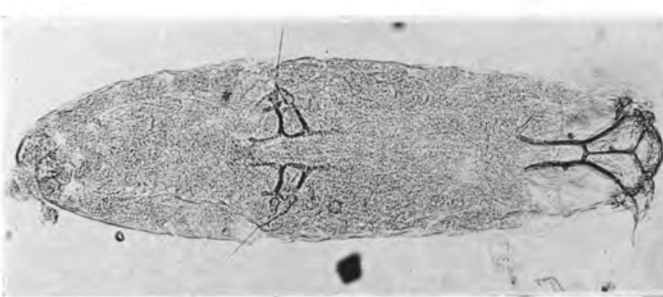

FIG. 249—**Falculifer rostratus,** nymph, from subcutis of a pigeon. x 60.

POULTRY

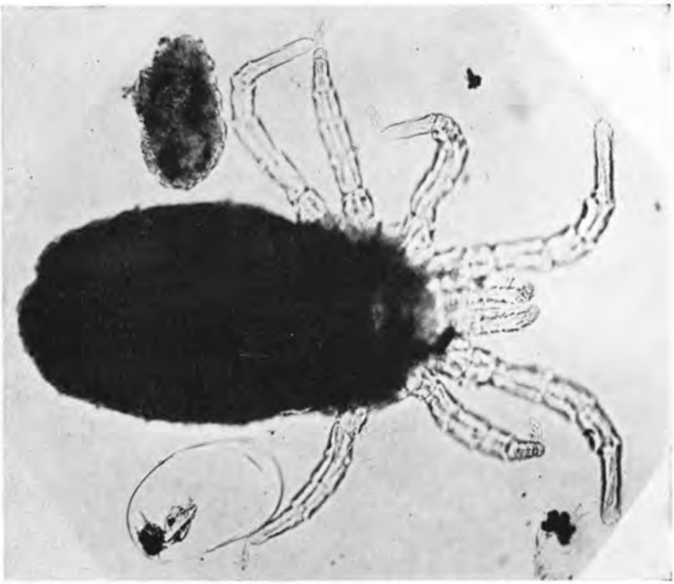

FIG. 250—Adult female **Dermanyssus gallinae,** the common red mite of poultry. x 65.

FIG. 251—Adult female **Bdellonyssus sylviarum,** the northern feather mite of poultry. x 75.

CATTLE

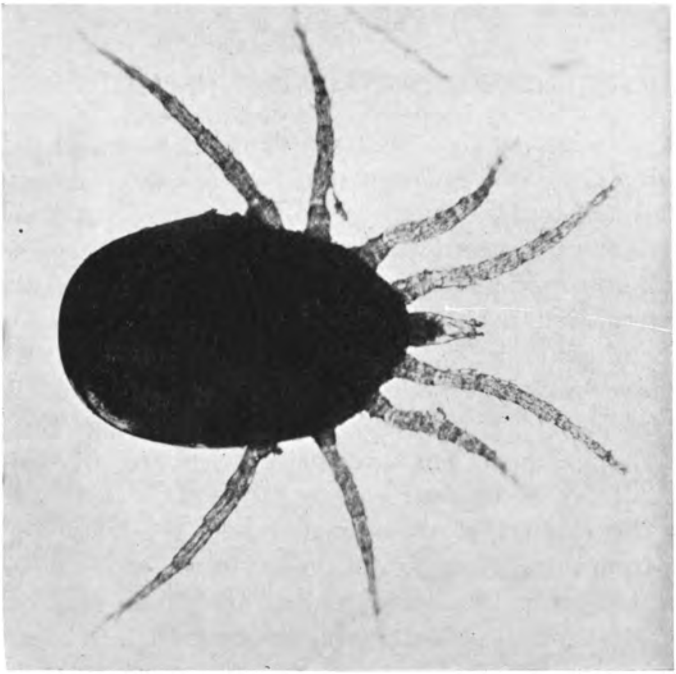

FIG. 252—Adult female **Raillietia auris,** a rarely reported ear mite of cattle. From the tympanic membrane of a steer at Ames, Iowa, March 10, 1925. x 35.

DOG

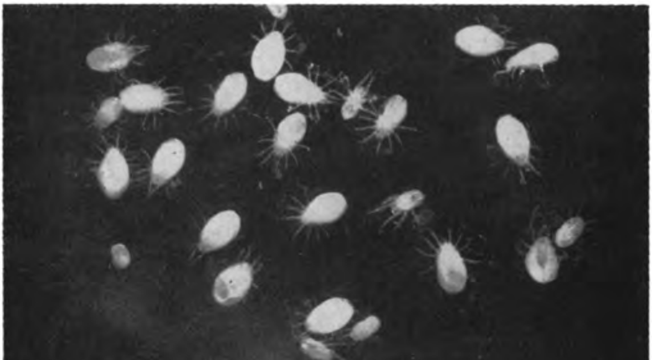

FIG. 253—**Pneumonyssus caninum,** the frontal sinus mite of dogs. Adults and larvae are seen; also an ovum at the lower left. Note the millimeter scale below the mites. x 7.

SECTION 3

The Diagnosis of Louse Infestations

Lice are wingless, dorso-ventrally flattened insects of the order Anoplura. They are important skin parasites of all domesticated mammals and birds. Lice are usually quite host-specific, that is, with few exceptions, each species of lice can live and reproduce on only one host species. The entire life cycle is spent on the host, and transmission is almost entirely by means of host contacts. The size of adult lice varies from slightly more than 1 mm. for the smaller species, to approximately 5 mm. in length for the larger species. Their bodies are distinctly divided into head, thorax, and abdomen. The three pairs of legs are attached to the thorax. All lice fasten their eggs (nits) to the hair of mammals and to the feathers of their avian hosts. The nymphs, which emerge from the eggs, are quite similar to the adults except that they are smaller, paler-colored, and do not possess mature sexual organs. Most species of lice complete a generation in about three weeks.

Technique for the Diagnosis of Lice Infestation

Most species of lice may easily be seen with the unaided eye. Louse eggs (nits) may likewise be observed, attached to the hair or feathers (Figs. 262, 265). Bird lice often attach their eggs in clusters at the feather bases (Fig. 271). Biting lice attract attention by their rapid movements. The examiner may acquire biting lice on his hands, arms, or body, especially if he handles the cadaver of a louse-infested animal several hours after death.

A hand lens of at least x 3 magnification is very helpful in the detection of lice and their eggs. If microscopic observation is desired, lice may be captured by means of a finely-pointed forceps, placed in a drop of water or mineral oil on a slide, and immobilized by means of a coverglass. Low power (x 100) is usually sufficient for the demonstration of morphologic details.

Lice are separated into two suborders, Mallophaga and Anoplura, depending upon feeding habits.

(1) *The Mallophaga.* These are the chewing or biting lice, so called because the anteriorly-rounded head is provided with mandible-like mouth parts (Fig. 270). They eat skin scales, feathers, skin secretions, and other organic debris found upon the skin. Certain of the bird lice apparently puncture the bases of the young quills, thus obtaining blood. It is quite probable that the biting lice will eat the blood that comes from skin wounds. In general, biting lice are yellow. Their legs are adapted for rapid movement over the skin and its coverings. All species of bird lice and the cat louse are of the biting type.

Species of chewing (biting) lice and their hosts:

Bovicola pilosa – Horse (Fig. 254)
Bovicola bovis. Red louse – Cattle (Fig. 256)
Bovicola ovis – Sheep (Fig. 260)
Bovicola peregrina – Sheep
Bovicola caprae – Goat
Bovicola limbata. Large yellow louse – Goat
Bovicola hermsi – Goat
Trichodectes canis – Dog, wolf (Fig. 266)
Trichodectes floridanus – Dog
Heterodoxus longitarsus. Marsupial louse – Dog, kangaroo, opossum (?) (Fig. 267)
Felicola subrostrata – Cat
Eomenacanthus stramineus. Body louse – Chicken, turkey (Figs. 269, 270)
Menopon gallinae. Shaft, or small body louse – Chicken, turkey, guinea fowl
Lipeurus heterographus. Head louse – Chicken
Lipeurus caponis. Wing louse – Chicken
Goniocotes gigas. Large louse – Chicken, guinea fowl
Goniocotes hologaster. Fluff louse – Chicken, guinea fowl
Goniodes dissimilis. Brown louse – Chicken
Lipeurus gallopavonis. Slender louse – Turkey
Goniodes meleagridis. Large louse – Turkey
Goniodes numidae. Feather louse – Guinea fowl
Lipeurus numidae. Slender louse – Guinea fowl
Anaticola crassicornis – Duck

Anatoecus dentatus — Duck, goose
Anaticola anseris. Slender louse — Goose
Trinoton anserinum. Body louse — Goose
Columbicola columbae. Slender louse — Pigeon
Goniocotes bidentatus. Small louse — Pigeon
Goniodes damnicornis. Little feather louse — Pigeon
Colpocephalum turbinatum. Narrow body louse — Pigeon

(2) *The Anoplura.* These include the suctorial lice. In general they are larger than the chewing lice, and are colored gray to dusky red, depending upon the amount of host's blood they contain. The head of the suctorial louse is elongated in order to accommodate the protrusible, piercing mouth parts. They are comparatively slow-moving insects, and are most frequently seen head down close to the skin surface. Their legs are adapted for firmly clasping the hair of the host. Suctorial lice are more pathogenic than the chewing lice because of their blood-sucking habits. All species of domesticated mammals, except cats and birds, harbor suctorial lice.

Species of suctorial lice and their hosts:

Haematopinus asini — Horse (Fig. 255)
Haematopinus eurysternus. Short-nosed louse — Cattle (Fig. 257)
Haematopinus quadripertussus. Tail louse — Cattle
Linognathus vituli. Long-nosed louse — Cattle (Fig. 258)
Solenopotes capillatus. Hairy, or little blue louse — Cattle (Fig. 259)
Linognathus pedalis. Foot louse — Sheep (Fig. 261)
Linognathus ovillus. Body louse — Sheep
Linognathus africanus. Blue louse — Goat, sheep
Linognathus stenopsis. Blue louse — Goat
Haematopinus suis. Common louse — Swine (Figs. 262 to 265)
Linognathus setosus — Dog, fox, coyote, ferret (Fig. 268)

HORSE

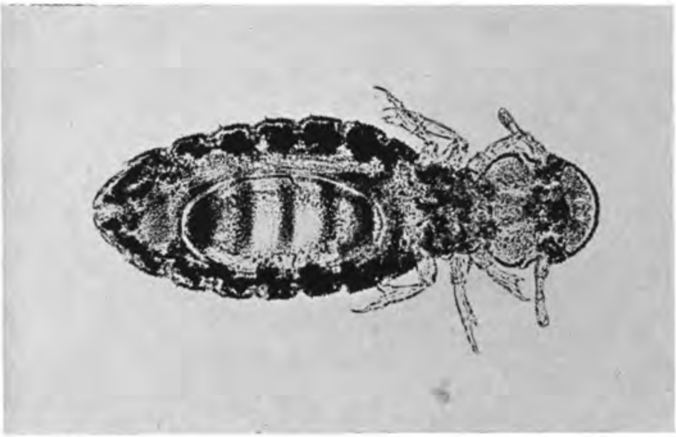

FIG. 254—Adult female **Bovicola pilosa,** the biting louse of horses. x 32.

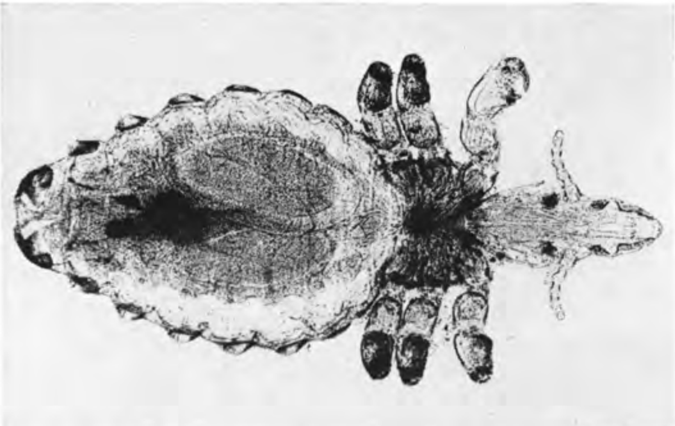

FIG. 255—Adult female **Haematopinus asini,** the suctorial louse of horses. x 25.

CATTLE

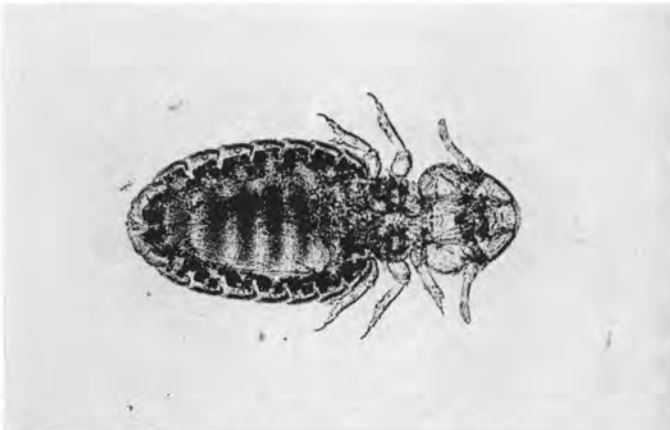

FIG. 256—Adult female **Bovicola bovis,** the biting louse of cattle. x 32.

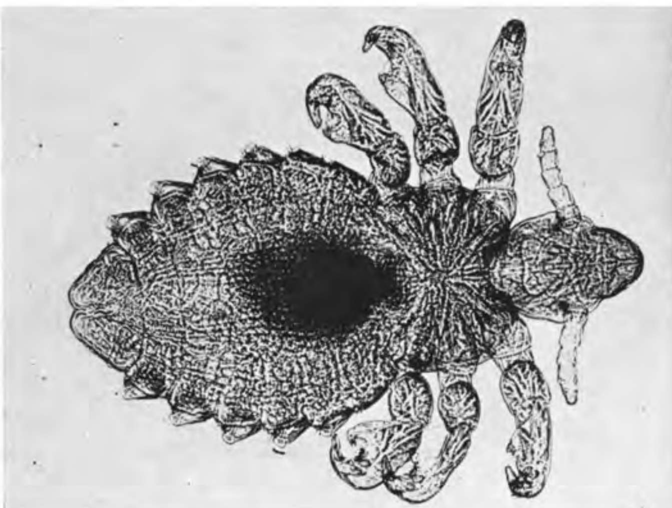

FIG. 257—Adult female **Haematopinus eurysternus,** the short-nosed suctorial louse of cattle. x 40.

CATTLE

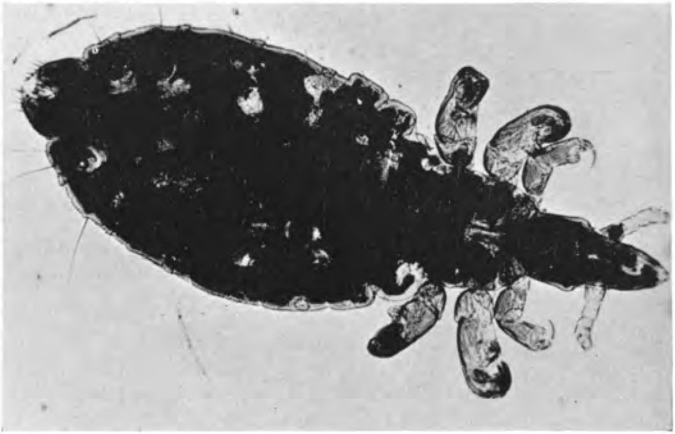

FIG. 258—Adult female **Linognathus vituli,** the long-nosed suctorial louse of cattle. x 40.

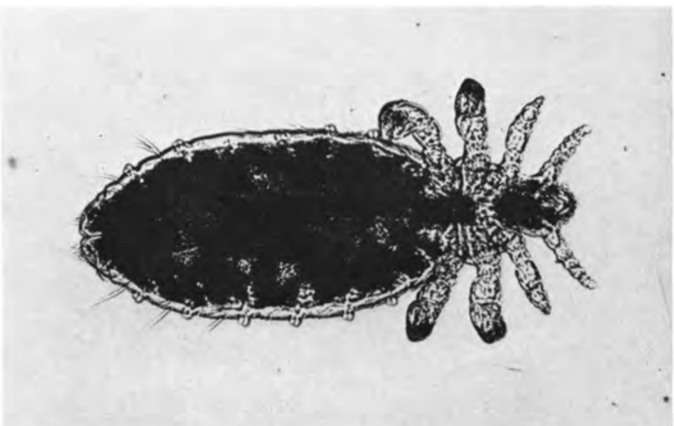

FIG. 259—Adult female **Solenopotes capillatus,** the little blue cattle louse. x 40.

SHEEP

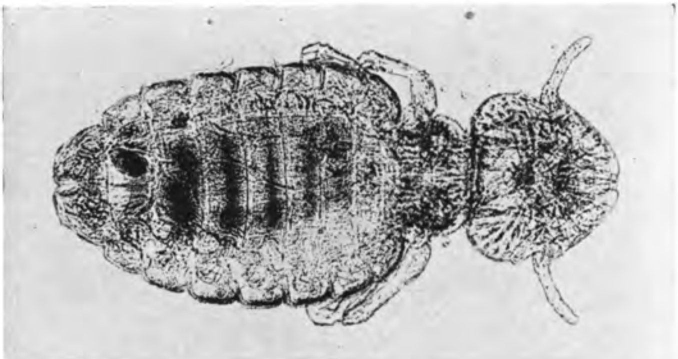

FIG. 260—Adult female **Bovicola ovis,** one of the species of biting lice of sheep. x 50.

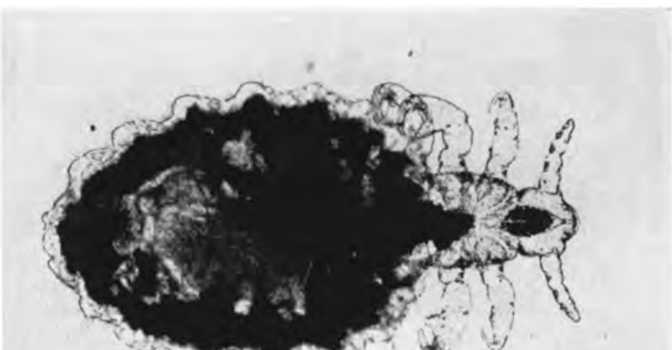

FIG. 261—Adult female **Linognathus pedalis,** the suctorial foot louse of sheep. x 37.

SWINE

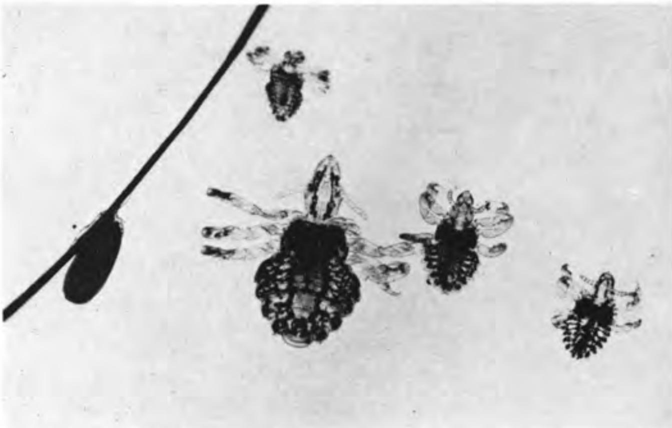

FIG. 262—Egg and nymphal stages of **Haematopinus suis,** the swine louse. x 10.

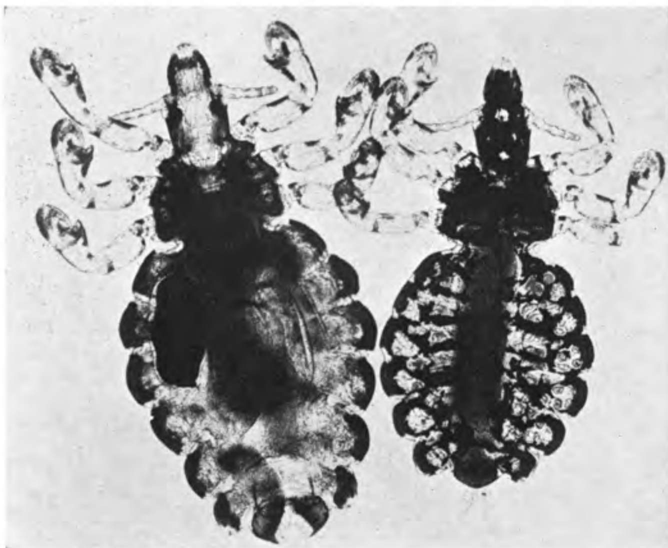

FIG. 263—Adult female **(left)** and male **(right)** swine lice, **Haematopinus suis.** x 15.

SWINE

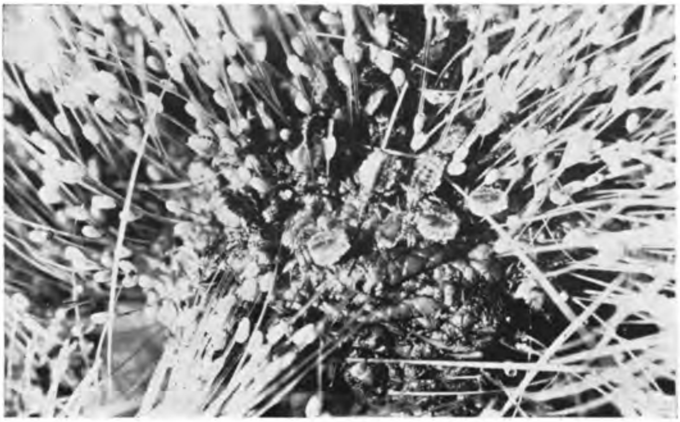

FIG. 264—Swine lice, **Haematopinus suis,** and their eggs on the skin. x 1.3.

FIG. 265—Eggs of **Haematopinus suis,** the swine louse, attached to hairs. x 2.

DOG

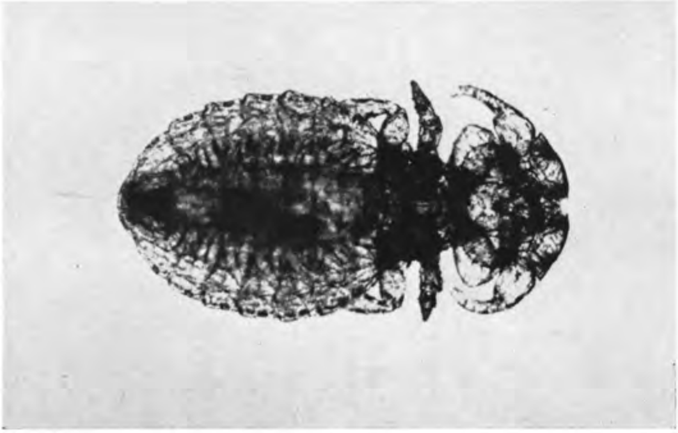

FIG. 266—Adult female **Trichodectes canis,** the common biting louse of dogs and wolves. x 35.

FIG. 267—Adult female **Heterodoxus longitarsus,** one of the biting lice of dogs, kangaroos, and probably opossums. x 40.

DOG

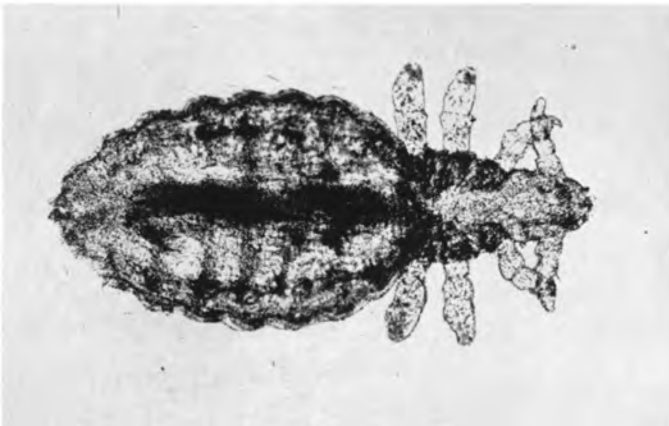

FIG. 268—Adult female **Linognathus setosus,** the suctorial louse of dogs, foxes, coyotes, and ferrets. x 40.

CHICKEN, TURKEY

FIG. 269—Adult female **Eomenacanthus stramineus**, the body louse of chickens and turkeys. x 25.

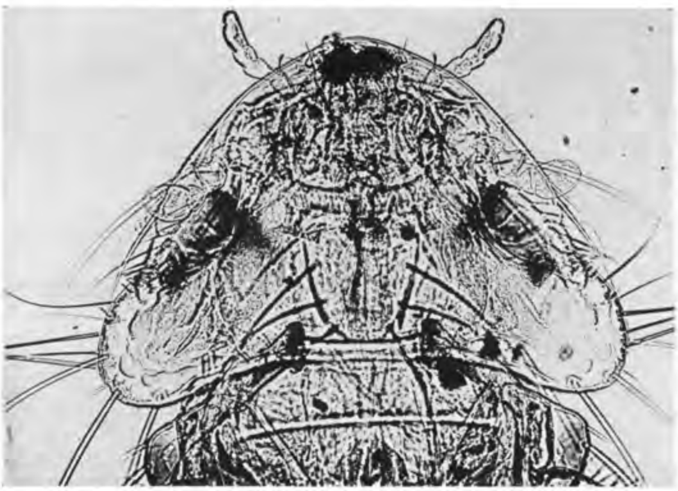

FIG. 270—Head of a biting louse, **Eomenacanthus stramineus**, the body louse of chickens and turkeys. x 100.

CHICKEN

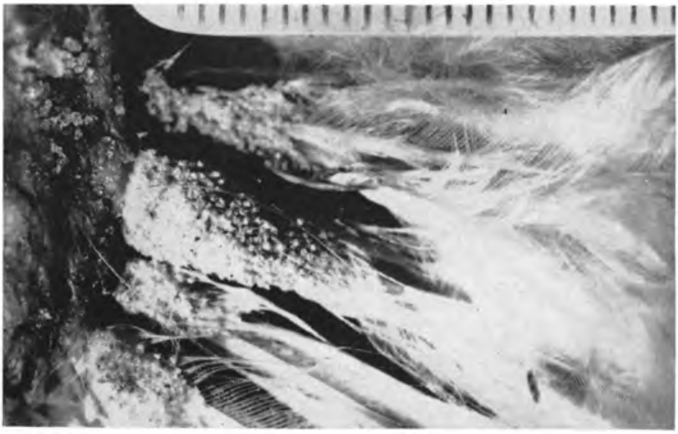

FIG. 271—Louse eggs on the bases of the feathers of a chicken. x 2.7.

References for Section 1

FECAL EXAMINATION IN THE DIAGNOSIS OF PARASITISM

Albert, H. 1927. Technic of examination of feces for amebas and other intestinal protozoa. Arch. Path. and Lab. Med. 3:235–37.

Alicata, J. E. 1935. (Comparative morphology of eggs and third-stage larvae of some nematodes occurring in swine.) *In*: U. S. D. A., Tech. Bul. 489:85–86.

———. 1941. Studies on the control of the liver fluke of cattle in the Hawaiian Islands. Amer. Jour. Vet. Res. 2:152–64.

Allen, J. A. 1922. The application of Vajda's method to the examination of fox feces. Jour. Am. Vet. Med. Assoc. 62 (n.s.15) :349–52.

Ando, A., and K. Kobori. 1926. On the results of the fecal examination of the dog and cat, in the region in Gifu-Prefecture thickly infested with paragonomiasis. Aichi Igakkwai Zasshi. 33 (4), July.

Andrews, Justin, C. M. Johnson, and S. C. Schwartz. 1934. The diagnosis of intestinal protozoa from purged and normally-passed stools. Jour. Parasit. 20: 253–54.

Andrews, M. M. 1935. The examination of faeces for the ova of *Schistosoma japonicum*. Chinese Med. Jour. 49:42–46.

Apo, H. 1926. Results of the examination of the soil and vegetables for the eggs of parasites. Tokyo Iji-Shinshi (2452), Jan.

Augustine, D. L., M. Nazmi, M. M. Helmy, and E. G. McGavran. 1928. The ova-parasite ratio for *Ancylostoma duodenale* and *Ascaris lumbricoides*. Jour. Parasit. 15:45–51.

Avinée, E., and J. L. Tiprez. 1930. Sur la régularité des pontes de *l'Ascaris* et du trichocéphale dans les feces. Compt. Rend. Soc. Biol., Paris, 102:1030–31.

Ayulo Robles, V. M. 1942. Metodos de diagnostico microscopio de la amebiasis. Rev. Med. Exp. (Lima, Peru), 1:177–94.

Bach, F. W. 1929. Leitfaden zur Untersuchung auf die parasitischen Protozoen des menschlichen Darm-Kanales. Jena, Germany. 140 pp.

Bacigalupo, J. 1931. La investigación de huevos de *Taenia saginata* en las materias fecales de los portadores del parásito. Semana Med. 38:1234–35.

———. 1936. Sobre le presencia accidental de huevos de helmintos en las materias fecales humanas. Su significatión clínica. Su diagnóstico. Rev. Parasit. Clín. y Lab., Habana, 2:267–76.

Bagby, B. B. 1907. A simple method of finding the ova of *Uncinaria*. Jour. Am. Med. Assoc. 48:325.

Bak, I. 1916. Bijdrage tot de diagnostiek der darmparasieten. Nederl. Tijdschr. Geneesk. 60, 2.R., 52, pt. 2 (13) :1117–22.

Bancroft, F. W., and E. S. Cross. 1906. Notes on the finding of *Coccidium bigeminum*. Johns Hopkins Hosp. Bul. 189, v.17:370–72.

Barker, L. F. 1916. Examination of the feces. Monogr. Med. 3:412–33.

———. 1916. Intestinal parasites, especially worms and their eggs. Monogr. Med. 3:433–59.

Barlow, C. H. 1931. A new method for examining urine for helminth eggs. Am. Jour. Hyg. 14:212–17.

Baroody, B. J. 1946. Modification of the Faust method in the detection of cysts and ova. Jour. Lab. and Clin. Med. 31:1372–74.

———. 1948. Comparative study of zinc sulfate and saline flotation methods in stool examination. Lab. diag. 12 (2) :9.

———, and H. Most. 1946. The relative efficacy of water centrifugal sedimentation and other methods of stool examinations for diagnosis of schistosomiasis japonica. Jour. Lab. and Clin. Med. 31:815–23.

Barthélemy, E. 1917. Essai de coprologie microscopique; diagnostic microscopique des maladies parasitaires à protozoaires et à helminthes. Paris, 128 pp.

Barto, L. R. 1936. Fecal analysis of the dog and a study of its importance in clinical diagnosis. No. Amer. Vet. 17:41–52 (July); 17:39–45 (Aug.).

Basnuevo, J. G., and V. Anido. 1936. La solucion aceto-formo-azucarado en el enriquecimiento de huevos de parásitos en las heces fecales. Med. Hoy. 1:49–50. (Abst. in Trop. Dis. Bul. 33:549–50.)

———, and ———. 1938. Conteo de huevos de helmintos. Rev. Med. Trop. y Parasitol. 4:85.

Bass, C. C. 1906. Uncinariasis in Mississippi. Jour. Am. Med. Assoc. 47:185–89.

———. 1909. Mild *Uncinaria* infection. Arch. Int. Med. 3:446–50.

———. 1910. The diagnosis of hookworm infection, with special reference to the examination of feces for eggs of intestinal parasites. Arch. Diag. 3:231–36.

Baughn, C. O., and A. Bliznick. 1954. The incidence of certain helminth parasites of the cat. Jour. Parasit. 40 (Sec.2) :19.

Beach, J. R. 1943. A rapid method for quantitative counts of coccidial oocysts in chicken feces. Cornell Vet. 33:308–10.

Beams, H. W. 1936. Survival of *Ascaris* eggs after centrifuging. Science, (n.s.84) :138.

de Beaurepaire Aragao, H. 1919. Novo metodo para facilitar o diagnostico e a conservação dos embryões de filarias no sangue e de parasitas nas fézes. Brazil-Med. 33:1–2.

Beaver, P. C. 1948. Progress report on a study of methods of pinworm diagnosis. Jour. Parasit. 34 (Sec.2) :21–22.

———. 1949. Quantitative hookworm diagnosis by direct smear. Jour. Parasit. 35:125–35.

———. 1949. A nephelometric method of calibrating the photo-electric light meter for making egg counts by direct fecal smear. Jour. Parasit. 35 (Sec.2) :13.

———. 1949. Methods of pinworm diagnosis. Amer. Jour. Trop. Med. 29:577–87.

———. 1950. The standardization of fecal smears for estimating egg production and worm burden. Jour. Parasit. 36:451–56.

Becker, E. R. 1926. Detection of intestinal protozoan infections by the cultivation method. Jour. Parasit. 12:219–20.

———, and W. Frye. 1927. Some protozoa found in the feces of cattle. Proc. Iowa Acad. Sci. 34:331–33.

———. 1934. (Notes on technique.) *In*: Coccidia and coccidiosis of domesticated, game and laboratory animals and of man. Collegiate Press, Inc., Ames, Iowa, pp. 121–22.

———. 1946. A survey of intestinal parasites in a unit of U. S. troops in Burma, with comments on methods for detecting the presence of *Entamoeba histolytica* in stools. Iowa Acad. Sci. 53:299–303.

Bekenskaia, A. I., L. D. Kashevnik, and V. A. Garibdzhanian. 1929. The question of helminthiasis in Leningrad and an appraisal of several methods used in examining faeces for the eggs of worms. (Russian text.) Vrach. Gaz. 33 (24), Dec. 31, col. 2815–17.

Bellama, R. 1932. Intestinal worms in Syria and Lebanon with special reference to *Ancylostoma duodenale*, analysis of one thousand stool examinations. Compt. Rend. Cong. Internat. Med. Trop. et Hyg. 4:301–5.

Benbrook, E. A. 1929. Fecal examination for evidence of parasitism in domestic animals. Jour. Am. Vet. Med. Assoc. 74 (n.s.27) :1009–26.

———. 1954. List of parasites of domesticated animals in North America. Burgess Pub. Co., Minneapolis, Minn. (Mimeographed), 67 pp.

Berndt, H. W. T. 1919. Vergleichende Stuhluntersuchungen auf Helmintheneier in Thüringen. Centralbl. f. Bakt. I Abt., Orig. 83:550–57.

Bidegaray, H. 1926. Nouvelle méthode de coloration pour l'examen microscopique des selles. Ann. de Parasit. 4:385.

———. 1927. Étude statistique et critique du parasitisme intestinal. La technique en coprologie. Thèse de médicine. Paris, 60 pp.

———. 1929. Sur un procédé pratique d'enrichissement en technique coprologique. Compt. Rend. Soc. Biol., Paris, 100:269–71.

Blerring, W. L., and H. Albert. 1903. Spurious tapeworms or parasitism due to undigested bananas. Iowa Med. Jour. 9:281–82.

Biester, H. E., and L. H. Schwarte (editors). 1952. Diseases of Poultry. Iowa State College Press, Ames, Iowa. 3rd ed., 1254 pp.
Biggam, A. G. 1930. The diagnosis of dysenteries. Jour. Roy. Army Med. Corps, 55:16–24.
Bijlmer, J. 1948. On the recovery of protozoa and eggs of some species of helminths in human feces. Jour. Parasit. 34:101–7.
Bingham, M. L. 1944. Some clinical diagnostic methods of use in conditions associated with animal parasites. Vet. Rec. 56:313–16.
Bittner, G. 1950. Detection of parasites in human excretions. Jour. Lab. and Clin. Med. 35:121–22.
Blackie, W. K., and W. A. McDonald. 1932. Eggs of Ternidens, Trichostrongylus, and Heterodera found in the faeces of man in Southern Rhodesia. Proc. Roy. Soc. Med. 25 (7), May, Sect. Trop. Dis. and Parasitol., p. 1067 (p. 27).
Blanchard, R. A. É. 1889. Pseudo-parasites. Dict. Encycl. Sc. Med., 2.s., 27:702–9.
———. 1898. Sur le pseudo-parasitisme des myriapodes chez l'homme. Arch. Parasit. 1:452–90.
———. 1902. Nouvelles observations sur le pseudo-parasitisme des myriapodes chez l'homme. Arch. Parasit. 6:245–56.
Blankmeyer, H. C. 1914. Intestinal myiasis with report of a case. Jour. Am. Med. Assoc. 63:321.
Blazhin, A. N. 1931. Beobachtungen der Eier von Parasitenwürmen auf einem dunkeln oder kontrastierenden Grund. Arch. f. Schiffs- u. Tropen-Hyg. 35:363–65.
Blount, W. P. 1941. Observations on the modified Gordon-Whitlock method for the counting of helminth ova in horse faeces. Jour. Roy. Army Vet. Corps, 12:69–78.
Boeck, W. C. 1917. A rapid method for the detection of protozoan cysts in mammalian faeces. Univ. Calif. Pub. in Zool. 18:145–49.
———. 1923. Technique of fecal examination for protozoan infections. Hyg. Lab., U. S. Pub. Health Serv. Bul. 133:62–74.
———, and C. W. Stiles. 1923. Studies on various intestinal parasites (especially Amoebae) of man. Hyg. Lab., U. S. Pub. Health Serv. Bul. 133:1–202.
Bogoiavlenskii, N. A., and A. Demidova. 1927. The value of the perianal scraper in the diagnosis of helminthiasis. (Russian text.) Russk. Zhurnal Trop. Med. 5:305–7.
———, and R. G. Lewitski. 1929. Würmträger unter den zur Wehrpflicht Einberufenen nach den Ergebnissen perianaler Abschabung. Arch. f. Schiffs-u. Tropen-Hyg. 33:413–16.
Böhm, L. K. 1920. Milben in den Faeces des Hundes; nebst Beiträgen zur Morphologie und Biologie der Milben. Wien. Tierärztl. Monatschr. 7:340–46; 361–72.
———. 1923. Morphologische und experimentelle Beiträge zur Kenntnis der Hunde- und Katzenkokzidiose. Wien. Tierärztl. Monatschr. 10:137–41.
———. 1928. Tierische Parasiten (in Harn). *In:* Stang u. Wirth, Tierheilk. u. Tierzucht, 22 (5):162–63.
———. 1928. (Kotuntersuchung auf Parasiten. Tierheilk. u. Tierzucht.) *In:* Stang u. Wirth. Lief. 27, v. 6, pp. 290–304.
———. 1933. Zum Artikel von H. Schereschewsky (Leningrad): "Bedeutung der Kotuntersuchungen zur Diagnostik der verschiedenen Räudearten bei Füchsen" in T. R. Jg. 38, Nr. 48, S. 381 (i.e. 831). Tierärztl. Rundsch. 39:65.
Bohrod, M. G. 1945. Detection of *Diphyllobothrium latum* ova in polarized light. Tech. Bul., Registry of Med. Technologists, 6:77–78.
Bomford, G. 1887. Note on eggs of *Distoma* (*Bilharzia*) *haematobium* found in transport cattle, Calcutta. Scient. Mem. Med. Off. India (1886), pt. 2:53–55.
Bonilla-Naar, A., and M. Gómez-Vargas. 1948. AEX and "Faust Simplificado" (Bonilla-Naar), two new methods for investigating intestinal parasitism. Jour. Parasit. 34 (Sec.2):32 (Abst.).
Borges Vierira, F., and G. Fleury da Silveira. 1930. Protozoarios encontrados nas fézes do homem na cidade de São Paulo. Brazil-Med. 44:1019.
Boston, L. N. 1901. Technique for the recognition of certain animal parasites in man. Am. Jour. Pharm. 73:228–33.

Boughton, D. C. 1937. Studies on oocyst production in avian coccidiosis. I. A dilution count technique. Am. Jour. Hyg. 25:187–202.
Bourne, R. F. 1938. The use of Babcock cream test bottles in the floatation of parasite eggs. Jour. Am. Vet. Med. Assoc. 93 (n.s. 46) :261–62.
Boycott, A. E., and J. S. Haldane. 1904. Ankylostomiasis, No. II. Jour. Hyg. 4:73–111.
Brand, A. F. A. 1931. Gastro-intestinal myiasis. Report of a case. Arch. Int. Med. 47:149–54.
Brandeis, R. 1909. Recherche des concrétions, des débris organisés, des parasites supérieurs dans les fèces. Dispositif pratique. Jour. Méd. Bordeaux. 39:743–44.
Braun, M., and M. Lühe. 1909. Leitfaden zur Untersuchung der Tierischen Parasiten des Menschen und der Haustiere für Studierende Ärzte und Tierärzte. Würzburg, 186 pp.
Briancon, L. 1904. De l'ankylostomiase et spécialement dans le bassin houiller de Saint-Étienne. Thèse méd. (Lyon), 353 pp.
Britton, J. W. 1937. Studies on the diagnosis of equine strongylidosis, with special reference to fecal and blood examinations. Cornell Vet. 27:290–96.
———. 1938. Rate of egg production of *Strongylus equinus* and *S. vulgaris* as measured by egg counts and qualitative larval cultures. Jour. Parasit. 24:517–20.
———. 1938. Studies on the normal variations in the strongyle egg counts of horse feces. Cornell Vet. 28:228–39.
Brooke, M. M., M. Goldman, and S. A. Johnson. 1949. Polyvinal alcohol fixative as a preservative and adhesive for protozoa in dysenteric stools and other liquid materials. Jour. Lab. and Clin. Med. 34:1554–60.
———, and B. R. Hogan. 1952. An evaluation of enteric parasitology performed in state laboratories. Pub. Health Rpts. 67:1237–48.
Broughton-Alcock, W., and J. G. Thomson. 1922. *Eimeria oxyspora* Dobell, 1919, found in a specimen of human faeces in England. Proc. Roy. Soc. Med. 15 (4), Feb., Sec. Trop. Dis. and Parasit.:1–7.
Brown, H. C., and G. E. F. Stammers. 1922. Observations on canine faeces on London pavements: bacteriological, helminthological and protozoological. Lancet 203 (v.2) :1165–67.
Brown, H. W. 1927. A study of the regularity of egg-production of *Ascaris lumbricoides, Necator americanus* and *Trichuris trichiura*. Jour. Parasit. 14:110–19.
———, and W. W. Cort. 1927. The egg production of *Ascaris lumbricoides*. Jour. Parasit. 14:88–90.
Brown, P. W. 1931. The diagnosis of endamebiasis. Med. Clin. No. Amer. 15:207–15.
Brown, R. L. 1945. Comparative studies on enterozoic parasite ova and cysts concentrating procedures. Am. Jour. Trop. Med. 25:375–76.
Brug, S. L. 1922. De parasitologische diagnostiek van de menschelijke faeces. Javasche Boekhandel en Drukkerij, Batavia, 75 pp.
———. 1936. Die Eosinmethode bei der Stuhluntersuchung auf Protozoen. Arch. f. Shiffs- u. Tropen-Hyg. 60:521–22.
Brumpt, E. J. A., and M. Neveu-Lemaire. 1933. Travaux pratiques de parasitologie. 2nd ed. Masson et Cie., Paris, 307 pp.
Bruns, H. 1914. Die mikroskopische Untersuchung der Fäzes in ihrer Bedeutung für die Bekämpfung der Ankylostomiasis. Ztschr. Hyg. u. Infektionskr. 78:385–416.
Brusaferro, D. 1887. Se dall' esame microscopico della feccie si possa dedurre la quantità dei distomi del fegato. Gior. Med. Vet. 36:296–304.
Bucki, A. J., and J. R. Vail. 1953. Technic for differentiating and preserving protozoa in feces. U. S. Armed Forces Med. Jour. 4:1195.
———, and W. H. Wells. 1953. A rapid technique for the diagnosis of intestinal protozoa. Science 117:235.
Burch, G. R., and F. A. Ehrenford. 1953. Canine strongyloidiasis. Vet. Med. 48:417–20.
Butler, R. L., and R. O. Christenson. 1942. A simple apparatus for determining the viability of embryonated helminth ova. Jour. Parasit. 28:131–34.

Caldwell, F. C. 1919. (Modification of the flotation loop method.) *In*: The Rockefeller Found. Ann. Rpt., p. 155.

———, and E. L. Caldwell. 1926. A dilution-floatation technic for counting hookworm ova in field surveys. Am. Jour. Hyg. 6 (March suppl.) :146–59.

Calkins, G. N. 1933. The biology of the *Protozoa*. 2nd ed. Lea and Febiger, Phila., Pa. 607 pp.

Cameron, T. W. M. 1934. (Egg techniques.) *In*: The parasites of domestic animals. A. and C. Black, Ltd., London, pp. 386–402.

———. 1942. (Technique.) *In*: The parasites of man in temperate climates. The Univ. Toronto Press, Toronto, pp. 170–73.

Campori, A. S. 1942. Diagnóstico de las teniasis en el perro por el método del hispo. Univ. Buenos Aires, Rev. Fac. Agron. y Vet. 9:170–80.

Camúñez, F. 1919. Una modificatión del método de Telemann para la investigación de huevos de parásitos intestinales. Actas Cong. Cien. Sevilla, vol. 9, Cien. Med. (1) :57.

Candela, M. 1923. Sui métodi di arrichimento per la ricerca delle uova de elminti nelle feci. Gazz. Internaz. Med. 28 (23) , Dec. 15:265–68.

Candlin, Frank. 1952. Improved fecal examination eliminates centrifuging. Auburn Vet., Winter 1952.

Carbone, D. 1911. Sulla ricerca delle uova elmintiche nelle fecce. Boll. Soc. Med.-Chir., Pavia. 25:261–63.

Carles, J., and E. Barthélemy. 1917. Procédé spécial d'homogénisation et detamisage pour collecter les kystes dysentériques contenus dans les selles. Compt. Rend. Soc. de Biol., Paris, 80:402–3.

Carne, H. R. 1927. On the preservation of rabbit faeces for transmission for examination as to the presence of oocysts of *Eimeria perforans* and *Eimeria stiedae*. N. S. Wales, Dept. Agr. Sci. Bul. 29:43–45.

Carter, H. F., and J. R. Matthews. 1917. The value of concentrating the cysts of protozoal parasites in examining the stools of dysenteric patients for pathogenic entamoebae. Ann. Trop. Med. and Parasit. 11:195–204.

Castellani, A., and A. J. Chalmers. 1920. Manual of tropical medicine. 3rd ed. Wm. Wood and Co., New York. 2439 pp.

Catcott, E. J. 1946. The incidence of intestinal protozoa in the dog. Jour. Am. Vet. Med. Assoc. 108:34–36.

Cauchemez, L. S. 1925. Technique et recherches de coprologie microscopique parasitaire chez le mouton et le porc. Thèse. Alfort, 78 pp.

Chandler, A. C. 1924. Animals as disseminators of hookworm eggs and larvae. Indian Med. Gaz. 59:533–37.

———. 1925. Notes on some methods for diagnosis of hookworm infection and for estimating the egg output. Indian Med. Gaz. 6:403–6.

———. 1929. (Diagnostic methods of fecal examination for eggs.) *In*: Hookworm disease. The Macmillan Co., New York, N. Y., pp. 437–52.

———. 1929. The weighted mean egg count as an index of the amount of hookworm infection in a community. Am. Jour. Hyg. 9:480–89.

———. 1955. Introduction to parasitology. John Wiley and Sons, Inc., New York, N. Y.

Chopra, R. N., and A. C. Chandler. 1928. (Synopsis of the commoner and more important tapeworm segments, nematode adults, nematode larvae and helminthic ova found in the faeces or other excreta or in the tissues of man and domestic animals.) *In*: Anthelmintics and their uses. Williams and Wilkins Co., Baltimore, Md., pp. 19–68.

Christensen, J. F. 1938. Species differentiation in the coccidia from the domestic sheep. Jour. Parasit. 24:453–67.

———. 1940. The effect of copper sulfate and ferric sulfate on coccidial oocyst output in feeder lambs. Jour. Am. Vet. Med. Assoc. 96:478–80.

———. 1941. The oocysts of coccidia from domestic cattle in Alabama (U. S. A.), with description of two new species. Jour. Parasit. 27:203–20.

Christenson, R. O. 1935. Remarques sur les différences qui existent entre les oeufs de *Capillaria aerophila* et de *Trichuris vulpis,* parasites du renard. Ann. Parasit. 13:318–21.

———, H. H. Earle, R. L. Butler, and H. H. Creel. 1942. Studies on the eggs of *Ascaridia galli* and *Heterakis gallinae*. Trans. Amer. Microsc. Soc. 61:191–205.

Christenson, R. O., L. Jacobs, F. G. Wallace, and M. B. Chitwood. 1940. (Nemic ova.) *In*: An introduction to nematology. Sec. I, Part III, Chap. XII. Leader Press. Babylon, N. Y., pp. 174–89.

Cilento, R. W., R. D. McIntosh, and N. B. Charlton. 1924. The diagnosis of bowel diseases in Northern Australia. Austral. Dept. Health Serv., Publ. 5, Melbourne, 84 pp.

Cobb, N. A. 1904. The sheep fluke. Fluke eggs as a quantitative aid in the diagnosis of the distomiasis of the sheep. Agr. Gaz., N. S. Wales, 15:658–69.

Cochran, S. 1915. Concentration of helminth ova from faeces. Chinese Med. Jour. 29:398–99.

Coffin, D. L. 1934. The diagnosis of lungworm disease in domestic ruminants, hogs and foxes by practical office-laboratory procedure. Univ. Pa., Vet. Exten. Quarterly, No. 90:41–46.

———. 1953. Manual of veterinary clinical pathology. 3rd ed. Comstock Pub. Co., Ithaca, N. Y. 322 pp.

Collier, W. A. 1927. Methoden zur Untersuchung parasitischer Würmer. Handbuch Biol. Arbeitsmeth. (Abderhalden.) Lief. 242, Abt. 9, Teil 1,2. Hälfte (4), pp. 661–92.

Cort, W. W. 1924. Investigations on the control of hookworm disease, XXXII. Methods of measuring human infestation. Am. Jour. Hyg. 4:213–21.

Craig, C. F. 1948. Laboratory diagnosis of protozoan diseases. Lea and Febiger, Phila., Pa. 384 pp.

———. 1944. The etiology, diagnosis and treatment of amebiasis. Williams and Wilkins Co., Baltimore, Md. 332 pp.

———, and E. C. Faust. 1951. (Aids for the identification of helminth eggs and larvae.) *In*: Clinical parasitology. 5th ed. Lea and Febiger, Phila., Pa., pp. 635–44.

Cram, E. B. 1925. The egg-producing capacity of *Ascaris lumbricoides*. Jour. Agr. Res. 30:977–83.

———. 1928. Spurious parasites from child and dog. (Banana fibre cells and citrus pulp vesicles.) Jour. Parasit. 14:202–3.

Crawley, H. 1921. Observations on the eggs of *Dictyocaulus filaria*. Jour. Am. Vet. Med. Assoc. 58 (n.s. 11):684–88.

———. 1925. Eggs of *Ankylostoma caninum*. Jour. Am. Vet. Med. Assoc. 66 (n.s. 19):487–89.

———. 1920. Eggs of *Toxascaris limbata*. Jour. Am. Vet. Med. Assoc. 69 (n.s. 22):493–97.

Cropper, J. W., and R. W. H. Row. 1916. A method of concentrating *Entamoeba* cysts in stools. Proc. Roy. Soc. Med. 10:1–12.

Crouch, H. B., and E. R. Becker. 1931. A method of staining the oocysts of coccidia. Science, 73:212–13.

Cushnie, G. H., and E. G. White. 1947. *Haemonchus contortus* infestation in lambs. Vet. Rec. 59:421–22.

———, and N. C. White. 1948. Seasonal variation in faeces worm-egg counts of sheep. Vet. Rec. 60:105–7.

D'Antoni, J. S., and V. Odom. 1938. A supplementary basic technique for the recovery of protozoan cysts and helminth eggs in feces. Preliminary communication. U. S. Pub. Health Serv., Pub. Health Rpts. 53:2202–4.

Darling, S. T. 1912. The examination of stools for the cysts of *Entamoeba tetragena*. Jour. Trop. Med. and Hyg. 15:257–59.

———. 1913. The identification of the pathogenic entamoeba of Panama. Ann. Trop. Med. and Parasit. 7:321–29.

———. 1915. Entamoebic dysentary in the dog. Proc. Assoc. Isthmian Canal Zone, 6:60–62.

Davaine, C. J. 1858. Sur le diagnostic de la présence des vers dans l'intestin par l'inspection microscopique des matières expulsées. Compt. Rend. Soc. Biol. (1857) 4:188.

Davis, N. C. 1924. Experience with the Stoll egg counting method in an area lightly infested with hookworms. Am. Jour. Hyg. 4:226–36.

Deem, A. W., and F. Thorp. 1939. Variation in numbers of coccidia in lambs during the feeding season. Vet. Med. 34:46–47.

Dennis, W. R., W. M. Stone, and L. E. Swanson. 1954. A new laboratory and field diagnostic test for fluke ova in feces. Jour. Am. Vet. Med. Assoc. 124:47–50.
de Rivas, D. 1928. An efficient and rapid method of concentration for the detection of ova and cysts of intestinal parasites. Am. Jour. Trop. Med. 8:63–72.
Deschiens, R. E. A. 1927. Comment dépister les infections parasitaires du tube digestif. Presse Méd. 35:3–4.
———, and R. Carvaillo. 1929. La coprologie en pratique médicale. Maloine, Paris, 129 pp.
Dikmans, G. 1945. Check list of the internal and external animal parasites of domestic animals in North America (United States and possessions, and Canada). Am. Jour. Vet. Res. 6:211–41.
di Primio, R. 1929. Do reconhecimento microscopico dos residuos fecaes de origen alimentar. Thèse de doctorat en médecine. Rio de Janeiro.
Dock, G., and C. C. Bass. 1910. Hookworm disease. Etiology, pathology, diagnosis, prognosis, prophylaxis and treatment. C. V. Mosby Co., St. Louis, Mo. 250 pp.
Doflein, F. J. T. 1953. Lehrbuch der Protozoenkunde. Aufl. 6. G. Fischer, Jena. 1190 pp.
Donaldson, R. 1917. An easy and rapid method of detecting protozoal cysts in faeces by means of wet-stained preparations. Lancet, 192:571–73.
Dumont, E. 1937, Beitrag zur Differential-diagnose parasitärer und nichtparasitärer Gebilde bei der mikroscopischen Untersuchung des Schweinekotes. Inaug. Diss., Tierärztl. Hochsch., Hannover, 24 pp.
Dunlap, W. R., P. A. Hawkins, and R. H. Nelson. 1949. Studies on sheep parasites. IX. The development of naturally acquired coccidial infections in lambs. Annals N. Y. Acad. Sci. 52:505–11.

Earle, W. C., and C. R. Doering. 1932. An evaluation of egg-count data in hookworm infestation. Amer. Jour. Hyg. 15:513–56.
Ehrenford, F. A. 1953. The incidence of some common canine intestinal parasites. Jour. Parasit. 39 (Supp., Sec. 2) :34.
———. 1954. Diagnosis of Physaloptera in dogs by stool examination. Jour. Parasit. 40 (Sec. 2) :16.
Ehrlich, K. 1927. Eine praktische Methode, Leberegeleier im Kote nachzuweisen. Tierärztl. Rundsch. 33:254–55.
Eigenfeld, D. D., and C. J. Schlesinger. 1944. An improved flotation method for the recovery of ova from feces. Jour. Am. Vet. Med. Assoc. 104:26.
Elsdon-Dew, R. 1947. Zinc sulphate flotation of faeces. Trans. Roy. Soc. Trop. Med. and Hyg. 41:213.
Emerson, C. P. 1910. The diagnostic importance of examination of the feces. Jour. Am. Med. Assoc. 54:270–75.
Emick, L. O. 1947. Statistical treatment of counts of trichostrongylid eggs. Biometrics Bul. 3:89–93.
Emrys-Roberts, E. 1915. Banana débris in faeces simulating tapeworm segments. Jour. Path. and Bact. 19:486–87.
Eyles, D. E., F. E. Jones, J. R. Jumper, and V. P. Drinnon. 1954. Amebic infections in dogs. Jour. Parasit. 40:163–66.

Farr, M. M., and R. W. Allen. 1942. Sulfaguanidine feeding as a control measure for cecal coccidiosis of chickens. Jour. Am. Vet. Med. Assoc. 100:47–51.
———, and G. W. Luttermoser. 1941. Comparative efficiency of zinc sulphate and sugar solutions for the simultaneous flotation of coccidial oocysts and helminth eggs. Jour. Parasit. 27:417–24.
Faust, E. C. 1924. Anomalies found in fecal examinations in China. China Med. Jour. 38:820–24.
———. 1949. (Examination of human excreta and body fluids for helminth eggs and larvae.) *In*: Human helminthology. 3rd ed. Lea and Febiger, Phila., Pa., 744 pp.
———. 1931. A study of canine amebic colitis. Porto Rico Jour. Pub. Health and Trop. Med. 6:391–400.
———. 1932. The symptómatology, diagnosis and treatment of *Strongyloides* infection. Jour. Am. Med. Assoc. 98:2276–77.

Faust, E. C. 1934. The distribution and diagnosis of amebic enteritis in the southern United States. New Orleans Med. and Surg. Jour. 86:605–9.

———, J. S. D'Antoni, V. Odom, M. J. Miller, C. Peres, W. Sawitz, L. F. Thomen, J. E. Tobie, and J. H. Walker. 1938. A critical study of clinical laboratory technics for the diagnosis of protozoan cysts and helminth eggs in feces. Am. Jour. Trop. Med. 18:169–83.

———, and W. A. Hoffman. 1934. Studies on schistosomiasis mansoni in Puerto Rico. III. Biological studies. 1. The extramammalian phases of the life cycle. Puerto Rico Jour. Pub. Health and Trop. Med. 10:1–97.

———, and J. W. Ingalls. 1946. The diagnosis of schistosomiasis japonica. III. Technics for the recovery of the eggs of *Schistosoma japonicum*. Am. Jour. Trop. Med. 26:559–84.

———, and O. Khaw. 1926. The egg-laying capacity of *Clonorchis sinensis*. Proc. Soc. Expt. Biol. and Med. 23:606–7.

———, and H. E. Meleney. 1924. Studies on schistosomiasis japonica. Am. Jour. Hyg., Monograph No. 3, 339 pp.

———, W. Sawitz, J. E. Tobie, V. Odom, C. Peres, and D. R. Lincicome. 1939. Comparative efficiency of various technics for the diagnosis of protozoa and helminths in feces. Jour. Parasit. 25:241–62.

———, W. H. Wright, D. B. McMullen, and G. W. Hunter III. 1946. The diagnosis of schistosomiasis japonica. I. The symptoms, signs and physical findings characteristic of schistosomiasis japonica at different stages in the development of the disease. Am. Jour. Trop. Med. 26:87–112.

Fehtkötter, H. 1926. Die Kochsalzmethode beim Nachweise von Wurmeiern, insbesondere bei Nebelkrähen. Vet. Med. Inaug. Diss., Berlin, 22 pp.

Felsenfeld, O. 1945. Comparison of methods used for the detection of *Endamoeba histolytica*. Jour. Parasit. 31 (Suppl.) :7.

———. 1950. Two survey methods used by public health laboratories for examination of stool specimens for Salmonellae, Shigellae and Protozoa. Pub. Health Rpts., Wash., D. C. 65:1075.

Fiebiger, J. 1947. Kurze Anleitung für das Untersuchen und Bestimmen der Parasiten. *In*: Die tierischen Parasiten der Haus- und Nutztiere, sowie des Menschen. Urban und Schwarzenberg, Berlin. 4 Aufl. 436 pp.

Fish, F. F. 1931. Quantitative and statistical analyses of infections with *Eimeria tenella* in the chicken. Am. Jour. Hyg. 14:560–76.

Fletcher, T. B. 1924. Intestinal Coleoptera. Indian Med. Gaz. 59:296–97.

Foster, W. D. 1912. Analysis of the results of 87 fecal examinations of sheep dogs for evidence of parasitism. Science, 35:553–54.

———. 1913. Some atypical forms of the eggs of *Ascaris lumbricoides*. Science, (n.s.37) :78.

———. 1914. Observations on the eggs of *Ascaris lumbricoides*. Jour. Parasit. 1:31–36.

Fouad, A. M. M. 1929. A new method for detecting *Schistosoma mansoni* ova in stools. Jour. Egypt Med. Assoc. 12:54.

Freeborn, S. B., and M. A. Stewart. 1937. (Dimensions of ova of nematodes attacking sheep.) *In*: Nematodes and certain other parasites of sheep. Calif. Agr. Exp. Sta., Bul. 603, pp. 7–8.

Freinkman, E. Z. 1930. Comparative evaluation of methods of fecal examinations in helminthiasis. Odesser med. Ztschr. 5:147.

Fülleborn, F. 1920. Neuere Methoden zum Nachweis von Helmintheneiern. Arch. f. Schiffs- und Tropen-Hyg. 24:174–76.

———. 1920. Die Anreicherungen der Helmintheneier mit Kochsalzlösung. Deutsche Med. Wchnsch. 46:714–15.

———. 1925. Eine Methode zur Isolierung von Hakenwurm- und anderen thermotaktischen-Larven aus Gemischen mit freilebenden Erdnematoden. Arch. f. Schiffs- und Tropen-Hyg. 29:470–78.

———. 1927. Zur "Hamburger Deckglasauszählung" für Hakenwurmeier. Arch. f. Shiffs- und Tropen-Hyg. 31:232–36.

Galli-Valerio, B. 1905. Notes de parasitologie et de technique parasitologique. Centralbl. f. Bakt. 1. Abt. Orig. 39:230–47.

———. 1919. Notes de parasitologie et technique parasitologique. Schweiz. Arch. f. Tierheilk. 61:289–302.

———. 1923. Parasitologische Untersuchungen und Beiträge zur parasitologischen Technik. Centralbl. Bakt. 1. Abt. Orig. 91:120–25.

———. 1932. Notes de parasitologie et de technique parasitologique. Centralbl. Bakt. 1. Abt. Orig. 125:129–42.

Galli-Valerio, B. 1933. Notes parasitologiques et de technique parasitologique. Centralbl. Bakt. 1. Abt. Orig. 129:422–33.

Garcia, E. Y., and T. P. Pesigan. 1940. The (IHP) centrifugal floatation method for the diagnosis of helminth ova and protozoan cysts in feces. Univ. Philippines Nat. and Appl. Sci. Bul. 7:299–303.

Garin, C. P., S. Doubrow, and H. Mounier. 1928. Les méthodes d'enrichissement appliquées à la recherche des oeufs de parasites dans les matières fécales. Notré modification de la méthode de Telemann. Lyon Méd. 141:341–45.

Garrison, P. E. 1910. Helminthological technique. 1. Methods for the collection, killing, preservation and mailing of parasitic worms and their ova. U. S. Naval Med. Bul. 4:345–54.

Gates, W. H. 1920. A method of concentration of parasitic eggs in feces. Jour. Parasit. 7:49.

Gelfand, M. 1948. The diagnosis of schistosomiasis in Southern Rhodesia by the rectal biopsy technique. Trans. Roy. Soc. Trop. Med. and Hyg. 42:283.

Giles, G. M. J. 1890. A report of an investigation into the causes of the diseases known in Assam as Kála-azár, and beri-beri. Shillong, 156 pp. (Staining technic for fecal examination.)

Gmelin, A. 1919. Vorkommen und Häufigkeit von Wurmeiern im Stuhl beobachtet an Verwundeten, Kranken und Angehörigen des Ldw.-Feld.-Laz. 33 und anderer Formationen. Centralbl. f. Bakt. 1. Abt. Orig. 83:460–66.

Goiffon, R. 1935. Manuel de coprologie clinique. 3rd ed. Masson et Cie., Paris, 274 pp.

———. 1945. Nécessité d'un régime sans résidus pour la recherche des oeufs et des kystes de parasites intestinaux. Arch. Malad. Appar. Digest. et Malad. Nutr. 34:341.

Goldman, M. 1947. Use of polyvinal alcohol to preserve fecal smears for subsequent staining. Science, 106:42.

———. 1948. Polyvinal alcohol-fixative method for shipping fecal smears. The Pub. Health Lab. 6:38–39.

———, and M. M. Brooke. 1953. Protozoa in stools: unpreserved and preserved in PVA-fixative. Pub. Health Rpts. 68:703–6.

———, S. A. Johnson, and Sadie A. Johnson. 1950. Deep-freeze preservation of stool specimens containing intestinal parasites. Jour. Parasit. 36:88.

Goldsby, A. I., and D. F. Eveleth. 1946. The diagnosis of internal parasites of sheep. Vet. Med. 41:398.

Gordon, H. M., and H. V. Whitlock. 1939. A new technic for counting nematode eggs in sheep faeces. Jour. Counc. Sci. and Indust. Res., Australia, 12:50–52.

Goss, L. W., and R. E. Rebrassier. 1922. Demonstration of the examination of the feces of the dog for parasitic infestation. North Am. Vet. 3:177–78.

Gould, S. E. 1945. (Laboratory diagnostic methods.) *In*: Trichinosis. Charles C Thomas, Springfield, Ill., pp. 165–88.

Gradwohl, R. B. H. 1948. (Examination of stools.) *In*: Clinical laboratory methods and diagnosis. 4th ed., 3 vols. C. V. Mosby Co., St. Louis, Mo.

Graham, C. F. 1941. A device for the diagnosis of Enterobius infection. Am. Jour. Trop. Med. 21:159–61.

Grand, A. 1927. Recherches de coprologie macro- et microscopique sur les débris alimentaires. Thèse de doctorat d'Université (Pharmacie), Paris.

Grassi, G. B., and E. Parona. 1879. Intorno all' anchilostomiasi, con un' appendice embriologica. Ann. Univ. Med., Milano v. 247, parte originale, 1. semestre, Magg., pp. 407–25.

Gromashevskii, L. V., and I. A. Shukhat. 1928. Mites in human faeces. Russk. Zhurnal Trop. Med. v. 6:209–16. German summary, pp. 278–79.

Guilford, H. G., and C. A. Herrick. 1952. Seasonal fluctuations in the number of coccidial oocysts and parasite eggs in the soil of pheasant shelter pens. Trans. Wisc. Acad. Sci. 41:153–62.

Guillaume A., and R. Noyer. 1929. Recherche coprologie de téguments de quelques graines de légumineuses; gesses, lupins et lentilles. Bul. Soc. Pharm. 36:345–50.

Habermann, R. T., F. P. Williams, Jr., and W. T. S. Thorp. 1954. Identification of some internal parasites of laboratory animals. U. S. Pub. Health Serv. Pub. No. 343, Natl. Inst. Health, 29 pp.

Hakansson, E. G. 1942. The use of aqueous smears in the examination of feces for intestinal protozoa. Am. Jour. Trop. Med. 22:325–27.

Haley, A. J. 1954. The use of a surface active agent to facilitate the examination of intestinal contents for helminth parasites. Jour. Parasit. 40:482.

Hall, M. C. 1912. A comparative study of methods of examining feces for evidences of parasitism. U. S. D. A., Bur. An. Indust., Bul. 135. 42 pp.

———. 1917. Apparatus for use in examining feces for evidences of parasitism. Jour. Lab. and Clin. Med. 2:347–53.

———. 1923. Diagnosis and treatment of internal parasites. Veterinary Medicine, Chicago, Ill. 2nd ed., 102 pp.

———. 1937. Studies on oxyuriasis. I. Types of anal swabs and scrapers, with a description of an improved type of swab. Am. Jour. Trop. Med. 17:445–53.

———, and D. L. Augustine. 1929. Some investigations of anthelmintics by an egg and worm count method. Am. Jour. Hyg. 9:584–628.

———, and E. B. Cram. 1925. Some laboratory methods for parasitological investigations. Jour. Agr. Res. 30:773–76.

Hausheer, W. C., and C. A. Herrick. 1926. The place of the smear in hookworm diagnosis. Am. Jour. Hyg. 6 (July suppl.) :136–48.

———, C. A. Herrick, and A. S. Pearse. 1926. Evaluation of the methods of Stoll and Lane in light hookworm infections, and accuracy in diagnosis of the Willis floatation method. Am. Jour. Hyg. 6:118–35.

Headlee, W. H. 1935. Studies on infections of human parasitic worms under institutional conditions. Jour. Lab. and Clin. Med. 20:1069–77.

———. 1942. Intestinal parasite infections among in-patients of the Indiana University Medical Center hospitals. Am. Jour. Trop. Med. 22:341–50.

Hegner, R. W. 1922. A comparative study of the giardias living in man, rabbit, and dog. Am. Jour. Hyg. 2:442–54.

———, and J. M. Andrews. 1930. Problems and methods of research in protozoölogy. The Macmillan Co., New York, N. Y. 532 pp.

———, W. W. Cort, and F. M. Root. 1923. Outlines of medical zoology. With special reference to laboratory and field diagnosis. The Macmillan Co., New York. 175 pp.

———, and L. C. Eskridge. 1935. Elimination and cross-infection experiments with trichomonads from fowls, rats, and man. Am. Jour. Hyg. 21:135–50.

———, F. M. Root, D. L. Augustine, and C. G. Huff. 1938. Parasitology, with special reference to man and domesticated animals. D. A. Appleton-Century Co., New York, N. Y. 812 pp.

———, and W. H. Taliaferro. 1924. Human protozoology. The Macmillan Co., New York, N. Y. 597 pp.

Heidegger, E. 1937. Wurmtafeln zum Bestimmen der wichtigsten Haustierparasiten. Ferdinand Enke, Stuttgart, 121 pp.

Hein, G. E. 1927. Cedar oil as an aid in finding parasite ova in feces. Jour. Lab. and Clin. Med. 12:1117–18.

Hellsten, H. 1933. Til fraagen om *Oxyuris'* paavisande, förekomst och betydelse. Nord. Med. Tidskr. 6:1358–63.

Hernández Morales, F., and J. Oliver González. 1945. Ova of *Schistosoma mansoni* in purged and unpurged fecal specimens. Puerto Rico Jour. Pub. Health and Trop. Med. 21:209–10.

Herrick, C. A. 1928. A quantitative study of infections with *Ancylostoma caninum* in dogs. Am. Jour. Hyg. 8:125–57.

Hetherington, D. C. 1922. Some new methods in nematode technic. Jour. Parasit. 9:102–4.
Heubner, O. 1922. Studien über Oxyuriasis. Jahrb. Kinderheilk. u. Phys. Erzieh. 98:1.
Hieronymi, E. 1925. Ueber Milbenbefunde im Stuhl. München. Med. Wchnschr., 72 (50), 11 Dez., p. 2517.
Hill, C. H., and R. E. Zimmerman. 1954. Use of a standard planetary-type mixing machine for separating eggs of the swine whipworm, *Trichuris suis,* from feces. Jour. Parasit. 40 (Sec.2) :32.
Hill, H. C. 1946. Observations on *Ancylostoma* and *Toxocara* infection in experimental and stock dogs. Jour. Parasit. 32:210.
Hill, R. B. 1923. Investigations on the control of hookworm disease. XXV. The use of the egg-counting method in an intensive campaign. Am. Jour. Hyg. 3 (July suppl.) :37–60.
———. 1926. The estimation of the number of hookworms harbored, by the use of the dilution egg count method. Am. Jour. Hyg. 6 (July suppl.) :19–41.
Hinman, E. H., and R. H. Kampmeier. 1934. Intestinal acariasis due to *Tyroglyphus longior* Gervais. Am. Jour. Trop. Med. 14:355–62.
Hirst, L. F. 1924. Investigations on the epidemiology of hookworm disease in Colombo. Part I. On the isolation and identification of infective nematode larvae. Part II. Observations on the viability of hookworm larvae. Ceylon Jour. Sci., Sect. *D.,* Med. Sci. 1:1–31.
Hitchcock, D. J. 1950. Parasitological study on the Eskimos in the Bethel area of Alaska. Jour. Parasit. 36:232–34.
———. 1951. Parasitological study on the Eskimos in the Kotzebue area of Alaska. Jour. Parasit. 37:309–11.
Hobmaier, M., and P. Taube. 1921. Die Kochsalzmethode bei Untersuchung auf Haustierparasiten. Berlin Tierärztl. Wchnschr. 37 (44) :521–22.
Hoffman, W. A., J. A. Pons, and J. G. Janer. 1934. The sedimentation-concentration method in schistosomiasis mansoni. Puerto Rico Jour. Pub. Health and Trop. Med. 9:280–90.
Howe, P. E., T. A. Rutherford, and P. B. Hawk. 1910. On the preservation of feces. Jour. Am. Chem. Soc. 32:1683–86.
Hung, S. L. 1926. Ueber den Nachweis von Hakenwurmeiern im Kote, den Wert ihrer quantitativen Bestimmung und eine einfache neue Methode für letztere. Arch. f. Schiffs- u. Tropen-Hyg. 30:399–421.
Hunter, G. W., L. S. Diamond, J. W. Ingalls, and E. P. Hodges. 1946. Studies on schistosomiasis; further studies on methods of recovering eggs of *S. japonicum* from stools. Anat. Rec. 96:515.
———, E. P. Hodges, W. G. Jahnes, L. S. Diamond, and J. W. Ingalls. 1948. Studies on schistosomiasis. II. Summary of further studies on methods of recovering eggs of *S. japonicum.* Bul. U. S. Army Med. Dept. 8:128–31.
———, J. W. Ingalls, and M. G. Cohen. 1945. Comparison of methods for diagnosing schistosomiasis japonica by recovery of eggs from feces. Jour. Parasit. (Suppl.) 31:21.
———, ———, and ———. 1946. Comparison of methods for recovery of eggs of *Schistosoma japonicum* from feces. Am. Jour. Clin. Path. 16:721–24.
Hussey, K. L., and N. E. Alger. 1951. Laboratory methods for the examination of mice for oxyurids. Jour. Parasit. 37:327.

Illinois University. 1939. Microscopic diagnosis of parasitism in domestic animals. (By Robert Graham *et al.*) Illinois Agr. Exp. Sta. Circ. 496. 123 pp.

Jackson, F. S. 1922. Note on the preparation of trematodes and nematodes for microscopic examination. Internat. Assoc. Med. Museums, Bul. 8:124–26.
Jacobs, A. H. 1942. Enterobiasis in children. Incidence, symptomatology, and diagnosis, with a simplified Scotch cellulose tape technique. Jour. Pediat. 21:497–503.
Jahnes, W. G., and E. P. Hodges. 1947. An improved method of sedimenting *Schistosoma japonicum* and other helminth ova. Jour. Parasit. 33:483–86.
Janssens, P. G. 1948. De "A.E.X." concentratie methode voor wormeneieren. Ann. Soc. Belg. Med. Trop. 28:213.

Jeffrey, G. M. 1950. Incidence of *Enterobius vermicularis* in Puerto Rican children, with a comparison of two diagnostic methods. Jour. Parasit. 36:485–88.
Jones, G. H. 1907. Examination of the feces. Therap. Gaz. 31, 3rd ser., 23:318–22.

Kalantarian, E. V. 1938. Utilisation du nitrate de sodium dans la pratique helminthologique. Med. Parazitol. i. Parazitar. Bolezni. 7:142–43. (Russian text, French summary.)
Kates, K. C. 1947. Diagnosis of gastrointestinal nematode parasitism of sheep by differential egg counts. Proc. Helminth. Soc. Wash. 14:44–53.
———, and D. A. Shorb. 1943. Identification of eggs of nematodes parasitic in domestic sheep. Am. Jour. Vet. Res. 4:54–60.
Kauzal, G. P., and H. M. Gordon. 1941. A useful mixing apparatus for the preparation of suspensions of faeces for helminthological examinations. Australia Counc. Sci. and Indust. Res. Jour. 14:304–5.
Keller, A. E. 1933. A study of the occurrence of unfertilized *Ascaris* eggs. Jour. Lab. and Clin. Med. 18:371–74.
———. 1934. A comparison of the efficiency of the Stoll egg-counting technique with the simple smear method in the diagnosis of hookworm. Am. Jour. Hyg. 20:307–16.
———. 1935. The occurrence of eggs of *Heterodera radicicola* in human feces. Jour. Lab. and Clin. Med. 20:390–92.
Kendrick, J. F. 1934. The length of life and rate of loss of the hookworms, *Ancylostoma duodenale* and *Necator americanus*. Am. Jour. Trop. Med. 14:363–79.
Kevorkova, V. I. 1946. (On the method of enterobiasis). Med. Parasitol. and Parasit. Dis. 15:73.
Khalil, M. 1921. On the occurrence of the eggs of mites in the faeces of miners in Cornwall and their subsequent development in culture media. Proc. Roy. Soc. Med. 14 (Trop. sect.) :24.
Kieffer, L. 1922. Die "Aufschwemm-Methoden" zum Nachweis der Parasiteneier im Kote der Haustiere. Inaug. Diss. (Giessen), 7 pp.
Knowles, R. 1928. An introduction to medical protozoology, with chapters on the spirochaetes and on laboratory methods. Thacker, Spink and Co., Calcutta. 887 pp.
Kofoid, C. A., and M. A. Barber. 1918. Rapid method for detection of ova of intestinal parasites in human stools. Jour. Am. Med. Assoc. 71:1557–61.
———, S. I. Kornhauser, and O. Swezy. 1919. Structure and relationships of the "iodine cysts" from human feces. Milit. Surg. 45:30–43.
Kolmer, J. A., and F. Boerner. 1945. Approved laboratory technic, clinical, pathological, bacteriological, mycological, parasitological, serological, biochemical, and histological. 4th ed. D. Appleton-Century Co., New York, N. Y. 1017 pp.
Kortenhaus, F. 1928. Verbesserter Nachweis von Wurmeiern im aufgehellten dicken Trockenkotausstrich. München. Med. Wchnschr. 75:1029.
Kouri, P., J. G. Basnuevo, and F. Sotolongo. 1943. (Diagnostico microscopico de las principales nematodiasis humanas.) *In*: Lecciones de parasitología y medicina tropical. Tomo II. Helmintología humana. Primera parte: Nemathelminthes. 2nd ed. La Habana, pp. 301–3.
———, ———, and ———. 1943. Lecciones de parasitología y medicina tropical. Tomo II. Helmintología humana. Primera parte: Nemathelminthes (vermes redondos.) 2nd ed. La Habana, 311 pp.
———, ———, and ———. 1944. Lecciones de parasitología y medicina tropical. Tomo II. Helmintología humana. Segunda parte: Plathelminthes (vermes planos). 2nd ed. La Habana, 336 pp.
Koutz, F. R. 1941. A comparison of floatation solutions in the detection of parasite ova in feces. Am. Jour. Vet. Res. 2:95–100.
———. 1944. Recent observations on parasites in small animals. Jour. Am. Vet. Med. Assoc. 104:199–203.
———, and R. E. Rebrassier. 1948. Identification and life cycles of parasites affecting domestic animals. Ohio State Univ. Press, Columbus, Ohio. 104 pp.
Kramer, F. 1925. Vergleichende Untersuchungen über den besten Nachweis von Parasiteneiern im Kot und einige Beobachtungen über die Entwicklung von *Ascaris mystax*. Deutsche Tierärztl. Wchnschr. 33:701–3.

Krug, E. S., and R. L. Mayhew. 1946. A comparative study of ova of four species of bovine gastro-intestinal nematodes. Jour. Parasit., Suppl., Sec. 2:17–18.

———, ———. 1949. Studies on bovine gastro-intestinal parasites. XIII. Species diagnosis of nematode infections by egg characteristics. Trans. Am. Micros. Soc. 68:234–39.

Krull, W. H. 1946. The identification of *Thysanosoma actinioides* infections in sheep by examination of fecal pellets. Trans. Am. Micros. Soc. 65:351–53.

Kudo, R. 1954. (Microscopical examination for protozoa.) *In*: Protozoology, 4th ed. Charles C Thomas, Springfield, Ill., 966 pp.

Kuenen, W. A., and N. H. Swellengrebel. 1913. Die Entamöben des Menschen und ihre praktische Bedeutung. Centralbl. f. Bakt. Orig. 71:378–410.

Kuitenen-Ekbaum, E. 1942. Diagnosis of enterobiasis: evaluation of recent devices. Can. Pub. Health Jour. 33:174.

Lane, C. 1918. Methods old and new for the detection of hookworm infection. Indian Jour. Med. Res. 6:1–25.

———. 1918. Preliminary note on an improved technique for the detection of hookworm eggs. Indian Med. Gaz. 53:173–74.

———. 1919. Diagnosis on a large scale in hookworm infection. Indian Jour. Med. Res., Special No.:186–204.

———. 1923–1927. The mass diagnosis of ankylostome infestation. Trans. Roy. Soc. Trop. Med. and Hyg. Part I, 16:274–315; Parts II to VII, 17:407–36; Parts VIII to XIII, 18:278-310; Part XIV, 19:156–76; Part XV, 20:455–77.

———. 1928. The mass diagnosis of hookworm infection. Am. Jour. Hyg. 8 (May suppl.) :1–48.

———. 1928. Recent advances in the diagnosis and treatment of human helminthiasis. Brit. Med. Jour. (3526) , Aug. 4:191–95.

———. 1932. Hookworm infection. Oxford Medical Publications, London, pp. 319.

———. 1933. Diagnosis of helminthic infections: a twelve-year survey. Trop. Dis. Bul. 30:503.

———. 1940. Hookworm diagnosis. Assumptions, alterations, controls, time-lag, rediscoveries: D.C.F. Trans. Roy. Soc. Trop. Med. and Hyg. 33:521–36.

Langeron, M. 1942. Précis de microscopie. Technique, experimentation, diagnostique. 6th ed. Masson et Cie., Paris, 1340 pp.

———, and M. Rondeau du Noyer. 1930. Coprologie microscopique. 2nd ed. Masson et Cie., Paris, 180 pp.

Law, R. G. 1930. (Hints on testing feces for the presence of parasites.) *In*: Feeding and diseases of the fox. Ontario Dept. Game and Fisheries, Bul. 2:29–30.

———. 1933. Hookworm infection in foxes. Ontario Dept. Game and Fisheries, Bul. 5. 33 pp.

Lawless, D. K. 1953. Rapid permanent-mount stain technic for diagnosis of intestinal protozoa: preliminary report. Am. Jour. Trop. Med. and Hyg. 2:1137.

Leichtenstern, O. 1886. Weitere Beiträge zur *Ankylostoma*-Frage. Deutsch. med. Wchnschr. 12 (11) , 18. März:173–76; (12) 25. März:194–96; (13) 1. Apr.:216–18; (14) 8. Apr.:237–40.

Letulle, M. 1905. Recherche des oeufs de parasites de l'intestin dans les matières fécales. Presse Méd., Paris, 13 (105) :841–43.

Levine, P. P. 1936. A new method for embryonating nematode eggs in fecal discharges. Jour. Parasit. 22:291.

Liebow, A. A., and C. A. Hannum. 1946. Eosinophilia, ancylostomiasis and strongyloidiasis in South Pacific area. Yale Jour. Biol. and Med. 18:381.

Lienhardt, H. F. 1930. Practical diagnosis of parasitism in animals. Vet. Bul. (Mimeographed Suppl. to Army Med. Bul.) , 24:220–22.

Liess, J. 1925. Vergleichende Untersuchungen über die Brauchbarkeit verschiedener Flotationsmedien zum Nachweis von Parasiteneiern im Kot der Haustiere. Inaug. Diss., Hannover, 64 pp.

Lopez-Neyra, C. R. 1953. Determinación de verminoses intestinales caninas, fase ineludible de toda lucha antihidatidica. Rev. Iberica Parasitol. 13:227–46.

Loughlin, E. H., and S. H. Spink. 1949. Diagnosis of helminthiasis. Jour. Am. Med. Assoc. 139:997.

Loughlin, E. H., and N. R. Stoll. 1946. An efficient concentration method (AEX) for detecting helminthic ova in feces (modification of the Telemann technic). Am. Jour. Trop. Med. 26:517–27.

Magath, T. B. 1916. Nematode technique. Trans. Am. Micr. Soc. 35:245–56.

———. 1934. The laboratory diagnosis of amebiasis. Jour. Am. Med. Assoc. 103:1218–24.

———, and C. B. Ward. 1928. Laboratory methods of diagnosing amebiasis. Am. Jour. Hyg. 8:840–57.

Maldonaldo, J. F., J. Acosta-Matienzo, and C. J. Thillet. 1949. Biological studies on miracidium of *Schistosoma mansoni*. II. Behavior of unhatched miracidium in undiluted stools under diverse environmental conditions. Puerto Rico Jour. Pub. Health and Trop. Med. 25:153.

———, ———, and ———. 1953. A comparison of fecal examination procedures in the diagnosis of schistosomiasis mansoni. Exp. Parasit. 2:294–310.

Manalang, C. 1928. Trichuriasis: relation between the number of ova per gram of formed stool and the number of females harbored by the host. Philippine Jour. Sci. 36:11–22.

Manson-Bahr, P., and W. J. Muggleton. 1945. Significance of mites and their eggs in human feces. Lancet, 1:81.

Maplestone, P. A. 1924. A critical examination of Stoll's method of counting hookworm eggs in faeces. Ann. Trop. Med. and Parasit. 18:189–94.

———. 1928. The rate of loss of hookworm eggs from feces. Indian Med. Gaz. 63:324–26.

———. 1929. A simple method of preserving faeces containing hookworm eggs. Indian Jour. Med. Res. 16:675–82.

Marsh, H. 1936. Observations based on weekly parasite egg counts on feces of lambs and yearling sheep. Jour. Parasit. 22:379–85.

———. 1938. Healthy cattle as carriers of coccidia. Jour. Am. Vet. Med. Assoc. 92 (n.s.45) :184–94.

Martins, A. V. 1936. Determination of *Schistosoma mansoni* in feces by means of concentration sedimentation. Brazil Med. 51:319–21.

Matuda, S. 1939. Some abnormal eggs of *Ascaris lumbricoides* L. Vol. Jub. pro Prof. Sadao Yoshida, 2:311–14.

May, H. G. 1922. On killing, staining and mounting nematodes. Jour. Parasit. 9:240.

Mazzotti, L. 1945. Presencia de huevecillos de varios helmintos, diferentes del *E. vermicularis,* en la region perianal de individuos examinados en Mexico para investigar oxyuriasis. Rev. d. Inst. Salub. y Enferm. Trop. 6:131.

———, and M. T. Osorio. 1945. The diagnosis of enterobiasis: comparative study of the Graham and Hall techniques in the diagnosis of enterobiasis. Jour. Lab. and Clin. Med. 30:1046–48.

———, L. Rodriguez, and A. Trevino. 1947. Observaciones en 161 personas parasitidas con Taenia. Rev. d. Inst. Salub. y Enferm. Trop. 8:155.

McCorkle, J. K. 1945. Modification of Faust-Meleney technic. Bul. U. S. Med. Dept. 45:420–22.

McDonald, J. D. 1920. Some limitations of the flotation method of fecal examination. Jour. Lab. and Clin. Med. 5:386–91.

McNeil, H. L. 1913. An improved method of extracting ova from stools. Jour. Am. Med. Assoc. 61:1628.

Meggitt, F. J. 1924. On the collection and examination of tapeworms. Parasitology, 16:266–68.

Mello, M. J., and R. Cuocolo. 1943. Tecnica para o xenodiagnóstico da habronemose gastrica dos equídeos. Arq. Inst. Biol. (Sao Paulo) 14:217–26.

Mhaskar, K. S. 1923. The diagnosis of hookworm infection. Indian Jour. Med. Res. 10:665–86.

Milaknis, A. 1953. An improved technic for fecal examination. Vet. Med. 48:41.

Miura, K., and N. Nishiuchi. 1902. Ueber befruchtete und unbefruchtete Ascarideneier im menschlichen Kote. Centralbl. Bakt. Parasit. 32:637–41.

Mönnig, H. O. 1928. Dilution egg counting on sheep faeces. Am. Jour. Hyg. 8:902–9.

———. 1947. (Clinical diagnostic methods.) *In*: Veterinary helminthology and entomology. 3rd ed. Williams and Wilkins Co., Baltimore, Md., pp. 16–29.

Morgan, B. B., and P. A. Hawkins. 1952. Diagnosis of protozoan infections. *In*: Veterinary protozoology. Burgess Pub. Co., Minneapolis, Minn., pp. 155–68.

———, and ———. 1949. Diagnosis of helminth infections. *In*: Veterinary Helminthology. Burgess Pub. Co., Minneapolis, Minn., pp. 345–64.

Morgan, D. O., and J. E. N. Sloan. 1947. Researches on helminths in hill sheep. With special reference to seasonal variations in worm egg output. Scot. Agr. 27:28–35.

Moss, E. S. 1938. Useful methods for routine and special examination of stools, with particular reference to the diagnosis of intestinal parasites. Am. Jour. Clin. Path., Tech. Suppl. 2:43–55.

Most, H. 1951. Diagnosis of intestinal helminths and protozoa: current perspective. *In*: Parasitic infections in man. Columbia Univ. Press, New York, pp. 56–75.

Nelson, E. C., and M. Bayliss. 1946. Schistosomiasis japonica, laboratory diagnosis. Bul. U. S. Army Med. Dept. 5:673–80.

Neveau-Lemaire, M. 1936. (Recherche et examen des oeufs d'helminthes.) *In*: Traité d'helminthologie médicale et vétérinaire. Vigot Frères, Paris, pp. 65–69.

———. 1943. (Recherche et étude des protozoaires parasites.) *In*: Traité de protozoologie médicale et vétérinaire. Vigot Frères, Paris, pp. 32–55.

Nickel, H. S. 1938. Hematoxylin staining of protozoan cysts obtained from feces by zinc sulphate levitation. Jour. Parasit. 24 (Suppl.) :8–9.

———. 1942. Amebiasis and hookworm infection as found in approximately 50,000 fecal examinations in Mississippi. Am. Jour. Trop. Med. 22:209–15.

Nissle, A., and O. Wagener. 1904. Zur Untersuchungstechnik von Eiern und Larven des *Ankylostomum duodenale*. Hyg. Rundschau, 14:57–60.

Nöller, W., and L. Otten. 1921. Die Kochsalzmethode bei der Untersuchung der Haustierkokzidien. Berliner tierärztl. Wchnschr. 37:481–83.

———, and F. Schmid. 1927. Die Wasserglas-Zentrifugier-Schwimm-Methode nach Vajda 1927 in ihrem Werte für parasitologische Untersuchungen. Tierärztl. Rundsch. 33:759–61.

Nomi, S. 1926. A staining method of goat coccidium and coccidium cyst. Jour. Jap. Soc. Vet. Sci. 5:94–96.

Obitz, K. 1934. Recherches sur les oeufs de quelques anoplocéphalidés. Ann. Parasit. 12:40–55.

Okoshi, S., S. Karasawa, Y. Hyuga, and H. Kubata. 1950. Research on methods of diagnosing bovine schistomiasis by means of rectal biopsies (trans. title). Jap. Jour. Vet. Sci. 12:281–83.

Olcinikow, S. W. 1929. Sur le diagnostic et l'épidémiologie dans l'enterobiose. (Russian text.) Russ. Jour. Trop. Med. 7:393–402. Abst. in Biol. Abst. 6:1123 (1933).

Oliver González, J., and F. Hernández Morales. 1946. Quantitative determinations of *Schistosoma mansoni* ova in feces from patients under treatment with antimonial drugs. Puerto Rico Jour. Pub. Health and Trop. Med. 22:210–16.

Olsen, O. W. 1946. Hexachlorethane-bentonite suspension for the removal of the common liver fluke, *Fasciola hepatica*, from sheep. Am. Jour. Vet. Res. 7:358–64. (New technic for fecal examinations.)

Otten, L. 1922. Die Kochsalzmethode bei der Untersuchung der Haustiercoccidien. Vet. med. Inaug. Diss., Berlin.

Otto, G. F., R. Hewitt, and D. E. Strahan. 1941. A simplified zinc sulfate levitation method of fecal examination for protozoan cysts and hookworm eggs. Am. Jour. Hyg. 33:32–37.

Ottolina, C. 1947. The rectoscopic biopsy by transparency: a new diagnostic method for *Schistosoma mansoni*. Am. Jour. Trop. Med. 27:603.

Payne, G. C., W. W. Cort, and W. A. Riley. 1923. Investigations on the control of hookworm disease. XX. Human infestation studies in Porto Rico by the egg-counting method. Am. Jour. Hyg. 3:315–38.

Pearse, A. S. 1942. (Worm eggs and examinations of feces for animal parasites.) *In*: Introduction to parasitology. Charles C Thomas, Springfield, Ill., pp. 173–86.

Pepper, W. 1908. A new method of examination of the feces for the ova of *Uncinaria*. With report of a case of *U. americana* and of a case of *U. duodenale*. Jour. Med. Res. 18 (n.s.13) :75–80.
Pesigan, T. P. 1940. Comparative efficiency of zinc sulphate and cupric nitrate technics for the diagnosis of helminth ova and protozoan cysts in feces. Univ. Philippines Nat. and Appl. Sci. Bul. 7:305–17.
Pfister, E. 1909. Die methodische Endoskopie (Proktoskopie) des bilharzia-kranken Enddarmes. Arch. f. Schiffs- u. Tropen-Hyg. 13:761–70.
Piana, G. P. 1906. Esame microscopico delle feci per la ricerca di elminti. La Clinica Vet. Anno. 29, n.1:15–20; 27–35; 49–55.
Pipkin, A. C. 1948. The diagnosis of taeniasis by perianal swab. Jour. Parasit. 34 (Sec.2) :27.
Pirot, R. 1924. La recherche des oeufs d'helminthes en coprologie. Etude expérimentale et critique des procédés d'enrichissement. Thèse de doctorat en médecine. Bordeaux.
Podyapolskaya, V. P. 1943. Diagnosis of helminthic infestations by the examination of scrapings from the perianal folds. Med. Parasit. and Parasit. Dis. 12:83.
Pusch, J., I. Senne, and W. Beyer. 1950. An improved, simple method for the identification of parasite eggs in fecal samples. Tierärztl. Umschau, 5:54.

Quadflieg, L. 1909. Ein Beitrag zur Faecesuntersuchung auf Parasiteneier. Deutsch. med. Wchnschr. 35 (48) :2106–7.

Ransom, B. H. 1906. The life history of the twisted wireworm (*Haemonchus contortus*) of sheep and other ruminants. U. S. D. A., Bur. An. Indus., Circ. 93. 7 pp.
Ratcliffe, H. L. 1944. A method for preparing permanent slides of the ova of parasitic worms. Science, 99:394.
Ray, D. K. 1953. Comparative efficiency of zinc sulfate flotation of coccidial cysts of sheep and goat. Proc. Zool. Soc. Bengal. 6:135–38.
Reardon, L. 1938. Studies on oxyuriasis. X. Artifacts in "cellophane" simulating pinworm ova. Am. Jour. Trop. Med. 18:427–31.
Rebrassier, R. E. 1940. Identification and life cycles of parasites affecting domestic animals. Ohio State Univ. Press, Columbus, Ohio, 89 pp.
———. 1940. Methods of diagnosis and treatment of the intestinal parasites of small animals. Cornell Vet. 30:133–40.
Rees, C. W. 1952. The processing of fecal specimens by the zinc sulfate flotation technique with safeguards for laboratory workers. Jour. Parasit. 38 (Sec.2) :26.
Regonesi, C., M. Muranda, and J. Artigas. 1954. Técnica del fijador con alcohol polyvinílico en el diagnóstico de la amibiasis y otras enteroparasitosis. Bol. Chileno de Parasitol. 9:105–9.
Reichenow, E., and G. Wülker. 1929. (Nachweis und Untersuchung von Wurmeiern.) *In*: Leitfaden der tierischen Parasiten des Menschen und der Haustiere. Curt Kabitzsch, Leipzig, pp. 126–37.
Reis, J., and P. Nobrega. 1936. (Diagnostico geral das helminthoses.) *In*: Tratado de doenças das aves. Inst. Biol., São Paulo, Brasil, pp. 348–51.
Riley, W. A., and R. O. Christenson. 1931. How to detect the parasites of fur-bearing animals. Univ. Minn., Agr. Ext. Div., Pamphlet 18. 22 pp.
Ritchie, L. S. 1948. An ether sedimentation technique for routine stool examinations. Bul. U. S. Army Med. Dept. 8:326.
———, C. Pan, and G. W. Hunter III. 1952. A comparison of the zinc sulfate and the MGL (formalin-ether) technics. Jour. Parasit. 38 (Sec.2) :16.
Rivera-Anaya, J. D., and J. Martinez de Jesús. 1951. Improved technique for the microscopic diagnosis of liver fluke infections in cattle. Jour. Agr., Univ. Puerto Rico, 35: (3) :98–99.
———, and ———. 1952. An improved technique for the microscopic diagnosis of liver fluke infection in cattle. Jour. Am. Vet. Med. Assoc. 120:203–4.
Roberts, F. H. S., and P. J. O'Sullivan. 1950. Methods for egg counts and larval cultures for strongyles infesting the gastro-intestinal tract of cattle. Austral. Jour. Agr. Res. 1:99–102.

Roberts, G. A. 1926. Interesting observations in the examination of feces of sucking calves, lambs and pigs (methods and findings). Jour. Am. Vet. Med. Assoc. 69 (n.s. 22) :75–79.
Robin, A. 1925. Sur une nouvelle méthode d'enrichissement des selles. Compt. Rend. Soc. de Biol. 92:1099.
Ross, I. C., and H. M. Gordon. 1936. (Diagnostic methods based on faecal examination and culture.) *In*: The internal parasites and parasitic diseases of sheep, their treatment and control. Angus and Robertson, Ltd., Sydney, Australia, pp. 195–208.
Royal, M. A. 1908. Intestinal pseudo parasites. Bul. State Univ. Iowa, n.s. 182.
Ruhe, D. S. 1940. Permanent mounts from NIH swabs positive for pinworm ova. Jour. Parasit. 26:333–34.
Ruotsalainen, A. 1911. Studier öfver förekomsten af tarmparasiter, speciellt *Oxyuris vermicularis,* hos barn. Finska läk.-sällsk handl. 53:444–66.
Rysavy, B., and B. Erherdova. 1952. A contribution to the diagnosis of helminthiasis of sheep and big game. (Eng. trans.) Zool. a Ent. Listy, 1:115–27.

Sarles, M. P. 1929. Quantitative studies on the dog and cat hookworm, *Ancylostoma braziliense,* with special emphasis on age resistance. Am. Jour. Hyg. 10, Sept. suppl.:453–75.
———. 1929. The effect of age and size of infestation on the egg production of the dog hookworm, *Ancylostoma caninum.* Am. Jour. Hyg. 10, Nov. suppl.:658–66.
Sasaki, R. 1927. On the oscillation of the specific gravity of the Ascaris eggs which are manifested by their growth. Tokyo Iji-Shinshi (Japanese text). English summary in Japan Med. World, 7:115.
Sawitz, W. 1942. The buoyancy of certain nematode eggs. Jour. Parasit. 28:95–102.
———, and E. C. Faust. 1942. The probability of detecting intestinal protozoa by successive stool examinations. Am. Jour. Trop. Med. 22:131–36.
———, V. Odom, and D. Lincicome. 1938. Comparative efficiency of the NIH anal swab examination and stool examination by brine and zinc sulphate floatation for *Enterobius* infection. Jour. Parasit. (Suppl.) 24:9.
———, J. E. Tobie, and G. Katz. 1939. The specific gravity of hookworm eggs. Am. Jour. Trop. Med. 19:171–79.
Schmid, F. 1944. Technik der Kotuntersuchung. *In*: Die parasitären Krankheiten unserer Haustiere. 4 Auflage. R. Schoetz, Berlin, pp. 41–60.
Schnelle, G. B., and T. C. Jones. 1944. Occurrence of the cereal mite in war dogs. Jour. Am. Vet. Med. Assoc. 104:213–14.
Schoop, G. 1925. Über die Diagnose der Entoparasiten des Schafes mittels der Kotuntersuchung. Inaug. Diss., Hannover.
Schroeder, K., and C. Jörgensen. 1914. Ueber das Vorkommen des *Trichocephalus dispar.* (Aus der med. Abt. Bdes. Krankenhauses zu Odense.) (Hospitalstidende 1913 Nr. 40 u. 41.) Muench. med. Wchnschr. 61 (5) :266.
Schuchmann, G., and L. Kieffer. 1922. Ueber den Nachweis von Parasiteneiern im Kote der Haustiere. Berl. Tierärztl. Wchnschr., Jahrg. 38, Nr. 19, S. 220–21; Nr. 31, S. 357–59.
Schüffner, W., and N. H. Swellengrebel. 1943. Methode zum Nachweis von Oxyuris Eiern: Ihre Leistung gegenuber dem amerikanischen NIH-Wischer. Zentralbl. f. Bakt., Parasit. u. Infekt. Krankh. 151:71.
Scott, J. A. 1937. The effect of various solutions on helminth eggs in feces. Jour. Parasit. 23:109–12.
———. 1937. Dilution egg counting in comparison with other methods for determining the incidence of *Schistosoma mansoni.* Am. Jour. Hyg. 25:546–65.
———. 1938. The regularity of egg output of helminth infestation with special reference to *Schistosoma mansoni.* Am. Jour. Hyg. 27:155–75.
———, and W. H. Headlee. 1938. Studies in Egypt on the correction of helminth egg count data for the size and consistency of stools. Am. Jour. Hyg. 27:176–95.
Seddon, H. R., and H. R. Carne. 1927. Incident of coccidiosis in Australian rabbits as determined by faecal examinations. N. S. Wales Dept. Agr., Sci. Bul. 29:33–41.

Seghetti, L. 1950. An improved method of mixing fecal suspensions for nematode egg counts. Proc. Hel. Soc. Wash. 17:26-27.

Seifried, O., and E. Heidegger. 1933. (Untersuchung auf Parasiten.) *In*: Pathologische Mikroskopie für Studierende und Tierärzte., pp. 16–17.

Serbinow, P. I., and E. S. Schulmann. 1927. Ueber die Methode der Analabschabung zur Oxyurisdiagnose. Arch f. Schiffs- u. Tropen-Hyg. 31:482–84.

Shakhnazarova, N. G. 1946. Helminthoscopic diagnosis of echinuriosis in ducks (trans. title). Proc. Moscow Zool. Park. 3:130–35.

Sheather, A. L. 1923. Detection of worm eggs in the faeces of animals and some experiences in the treatment of parasitic gastritis in cattle. Jour. Comp. Path. and Thera. 36:71–90.

———. 1923. The detection of intestinal protozoa and mange parasites by a floatation technic. Jour. Comp. Path. and Thera. 36:266–75.

———. 1924. The detection of worm eggs and Protozoa in the faeces of animals. Vet. Rec. 4:552–57.

Shillinger, J. E. 1937. Detecting internal parasites. *In*: Diseases of fur animals. U. S. Dept. Agr., Farmers' Bul. 1777:16–17.

Shorb, D. A. 1939. Differentiation of eggs of various genera of nematodes parasitic in domestic ruminants in the United States. U. S. D. A., Tech. Bul. 694. 10 pp.

———. 1940. A comparative study of the eggs of various species of nematodes parasitic in domestic ruminants. Jour. Parasit. 26:223–31.

Sigalas, R., and R. Pirot. 1924. Un nouveau procédé d'enrichissement des selles en coprologie. Compt. Rend. Soc. Biol. 90:755–57.

———, and A. Robin. 1925. Technique d'enrichissement des selles. Destruction des débris végétaux. Gaz. Hebd. Soc. Méd., Bordeaux. 17 Mai, No. 20.

Silva Leitão, J. L., J. M. Lino de Sousa, and L. D. Borges Ferreira. 1945. Método de Szepeshelyi e Urbany, no diagnóstico coprológico da distomatose ovina. Repositório de Trabalhos do Lab. Centr. de Path. Vet. 6 (1) :159–64 (Lisbon).

Simon, C. E. 1922. (Microscopic examination of the feces.) *In*: A manual of clinical diagnosis. 10 ed., Phila., Pa., pp. 361–73.

Skriabine, K. I., and E. E. S. Schulz. 1936. Les helminthoses pulmonaires des animaux. Bul. Off. Internat. Epizoot. 12:407–56. Abst. in Vet. Bul. 7:337–38 (1937).

Sloss, M. W. 1939. Spurious parasites in a dog. (Sheep parasite ova and oocysts.) The Vet. Student, 1:54.

Sluiter, C. Th., and N. H. Swellengrebel. 1912. De dierlijke parasiten van den mensch en van ouze huisdieren. 2. Aufl. Scheltema en Holkema, Amsterdam.

Smillie, W. G. 1921. A comparison of the number of hookworm ova in the stool with the actual number of hookworms harbored by the individual. Am Jour. Trop. Med. 1:389–95.

Snijders, E. P. 1920. On the cysts of a hitherto undescribed species of *Eimeria* in human stools. Parasitol. 12:427–32.

Soper, F. L. 1926. Comparison of the Stoll and Lane egg-count methods for the estimation of hookworm infestation. Am. Jour. Hyg. 6 (July suppl.) :62–102.

———. 1927. The relative egg-laying function of *Necator americanus* and *Ancylostoma duodenale*. Am. Jour. Hyg. 7:542–60.

Souegas, R. 1932. (Fecal technic for physicians and veterinarians.) *In*: Analyse micrographique. 2e. ed. Vigot Frères, Paris.

Southwell, T., and A. Kirshner. 1938. (Drawings of helminth eggs.) *In*: Guide to veterinary parasitology and entomology. H. K. Lewis and Co., Ltd., London, pp. 124–25.

Spedding, C. R. W. 1952. Variations in the egg count of sheep faeces within one day. Jour. Helminth. 26:71–86.

Spindler, L. A. 1928. Use of the egg isolation technic in epidemiological studies on ascariasis in Virginia. Jour. Parasit. 15:147.

———. 1929. On the use of a method for the isolation of ascaris eggs from the soil. Am. Jour. Hyg. 10:157–64.

Steck, W. 1926. A simple direct method for determining the number of worm ova in faeces. Schweiz. Arch. f. Tierhlk. 68:561–63.

Steel, E. R. 1930. Routine microscopic fecal examinations in small-animal practice. Jour. Am. Vet. Med. Assoc. 77 (n.s. 30) :9–17.
Stevenson, R. T. 1942. Use of calcium chloride to isolate helminth ova from soil. Jour. Parasit. 28 (Suppl.) :24.
Stiles, C. W. 1902. The significance of the recent American cases of hookworm disease (uncinariasis or anchylostomiasis) in man. 18th Ann. Rpt., Bur. An. Indust., U. S. D. A., pp. 183–219.
———. 1903. Clinical diagnosis of intestinal parasites. Jour. Am. Med. Assoc. 41:172–73.
———, and C. H. Gardner. 1910. On testing the viability of hookworm eggs. U. S. Pub. Health Serv., Pub. Health Rpts. 25:1825–30.
———, and A. Hassall. 1902. Spurious parasitism due to partially digested bananas. *In*: Eleven miscellaneous papers on animal parasites. U. S. D. A., Bur. An. Indust., Bul. 35:56–57.
Stitt, E. R., P. W. Clough, and M. C. Clough. 1945. (Fecal technic.) *In*: Practical bacteriology, haematology, and animal parasitology. 9th ed. P. Blakiston's Son and Co., Phila., Pa. 961 pp.
Stoll, N. R. 1923. Method for determination of number of hookworm eggs in given sample of feces. Jour. Parasit. 9:236.
———. 1923. Investigations on the control of hookworm disease. XIV. An effective method of counting hookworm eggs in feces. Am. Jour. Hyg. 3:59–70.
———. 1923. Investigations on the control of hookworm disease. XVIII. On the relation between the number of eggs found in human feces and the number of hookworms in the host. Am. Jour. Hyg. 3:156–79.
———. 1923. Investigations on the control of hookworm disease. XXI. On the use of an egg counting method in soil-culture studies of hookworm larvae. (Prelim. rpt.) Am. Jour. Hyg. 3:339–42.
———. 1924. Investigations on the control of hookworm disease. XXXIII. The significance of egg count data in *Necator americanus* infestations. Am. Jour. Hyg. 4:466–500.
———. 1929. Studies on hookworm, ascaris, and trichuris in Panama. II. Laboratory methods in the Panama studies. Am. Jour. Hyg., Monogr. Series, No. 9:30–44.
———. 1929. Studies on hookworm, ascaris, and trichuris in Panama. III. Stool size and its relation to eggs in the feces. Am. Jour. Hyg., Monogr. Series, No. 9:45–53.
———. 1930. On methods of counting nematode ova in sheep dung. Parasitol. 22:116–36.
———. 1946. *Necator americanus* and *Ancylostoma duodenale* in Guam, Leyte, and Okinawa, with a note on hookworm egg sizes. Jour. Parasit. 32:490–96.
———. 1948. El problema de la investigación de la uncinaria. Med. Revista Amer. 28:197.
———, W. W. Cort, and W. S. Kwei. 1927. Egg-worm correlations in cases of *Fasciolopsis buski*. Jour. Parasit. 13:166–72.
———, and W. C. Hausheer. 1926. Accuracy in the dilution egg-counting method. Am. Jour. Hyg. 6 (March suppl.) :80–133.
———, and ———. 1926. Concerning two options in dilution egg-counting: Small drop and displacement. Am. Jour. Hyg. 6 (March suppl.) :134–45.
———, and H. W. Tseng. 1925. The severity of hookworm disease in a Chinese group as tested by hemoglobin reading for the anemia and egg counts for the degree of infestation. Am. Jour. Hyg. 5:536–52.
Strileiue, D. 1932. Mikrophotographischer Atlas der Zerealien. Zeitschr. f. d. ges. Getreide u. Mühlenwesen. Berlin.
Stubbendorff, G. 1893. Die Differentialdiagnose der tierischen Parasiten-Eier und pflanzlicher Sporen. Inaug. Diss., Rostock. 33 pp.
Sullivan, B. H. 1954. Rectal biopsy in the diagnosis of schistosomiasis. Amer. Pract. 5:97–98.
Summers, W. A. 1942. Intestinal parasites in boys of the Florida Industrial School. Jour. Parasit. 28:169–70.
———. 1942. A modification of zinc sulphate centrifugal flotation method for recovery of helminth ova in formalinized feces. Jour. Parasit. 28:345–46.

Swales, W. E. 1939. Notes on the diagnosis and treatment of parasitic diseases of sheep in Canada. Can. Jour. Comp. Med. 3:341–44.

Swanson, L. E., and E. G. Batte. 1952. Internal parasites of cattle and their control. Vet. Med. 47:172–74; 176.

———, and H. H. Hopper. 1950. Diagnosis of liver fluke infection in cattle. Jour. Am. Vet. Med. Assoc. 117:127–29.

Swartzwelder, J. C. 1939. A comparison of five laboratory techniques for the demonstration of intestinal parasites. Jour. Trop. Med. and Hyg. 42:185–87.

———, and S. J. Cali. 1942. Human intestinal myiasis due to syrphid larvae. Report of an additional case (*Eristalis tenax*). Am. Jour. Trop. Med. 22:159–63.

Sweet, W. C. 1925. Average egg count per gram per female hookworm in Ceylon. Jour. Parasit. 12:39–42.

———. 1925. Notes on methods of diagnosing hookworm infection and on egg-counting methods. Am. Jour. Hyg. 5:497–507.

Szepeshelyi, A., and L. Urbany. 1934. Un método d'arricchemento per l'accertamento delle uova di destoma nelle feci (Münch. Tier. Woch. No. 25, Junho, 1934). La Clinica Vet.:955.

Taube, P. 1922. Eine Durchsuchung der Säugetiere des Zoologischen Gartens zu Berlin auf Wurmeier nach der Kochsalz-Methode. Vet. med. Inaug. Diss., Berlin.

Taylor, E. L. 1934. A method of estimating the number of worms present in the fourth stomach and small intestine of sheep and cattle for the definite diagnosis of parasitic gastritis. Vet. Rec. 14:474–76.

———. 1935. Seasonal fluctuation in the number of eggs of trichostrongylid worms in the faeces of ewes. Jour. Parasit. 21:175–79.

———. 1939. Diagnosis of helminthiasis by means of egg counts, with special reference to red-worm disease in horses. Vet. Rec. 51:895–98.

Telemann, W. 1908. Eine Methode zur Erleichterung der Auffindung von Parasiteneiern in den Faeces. Deutsch. Med. Wchnschr. 34:1510–11.

Tetley, J. H. 1941. *Haemonchus contortus* eggs: comparison of those in utero with those recovered from feces, and a statistical method for identifying *H. contortus* eggs in mixed infections. Jour. Parasit. 27:453–63.

———. 1941. The differentiation of eggs of the trichostrongylid species *Nematodirus filicollis* and *N. spathiger*. Jour. Parasit. 27:473–80.

———. 1949. Rhythms in nematode parasitism of sheep. New Zealand Dept. Sci. and Indust. Res., Bul. 96, 214 pp.

Theodorides, J. 1948. Les coléoptères, parasites accidentels de l'homme. Ann. Parasitol. Hum. et Comp. 25:348–63.

Thienel, M. 1925. Neue erfolgreiche Versuche zur Bekämpfung der Lebergelseuche. Münch. Tierärztl. Wochenschr., Jhrg. 76, Nr. 29, S. 621–32.

Thomson, J. G., and A. Robertson. 1926. Fish as the source of certain coccidia recently described as intestinal parasites of man. Brit. Med. Jour. 1:282.

Tobie, J. E., L. V. Reardon, J. Bozicevich, Bao-Chih Shih, N. Mantel, and E. H. Thomas. 1951. The efficiency of the zinc sulfate technic in the detection of intestinal protozoa by successive stool examinations. Ann. Trop. Med. 31:552–60.

Todd, A. C., F. E. Hull, G. W. Kelley, Z. N. Wyant, and M. F. Hansen. 1949. Worm parasites in thoroughbred mares. Ky. Agr. Exp. Sta. Bul. 536, 16 pp.

Todd, J. C., and A. H. Sanford. 1948. (The feces. Animal parasites.) *In*: Clinical diagnosis by laboratory methods. 11th ed. W. B. Saunders Co., Phila., Pa., pp. 479–603.

Tomb, J. N., and M. M. Helmy. 1931. Diagnosis of intestinal schistosomiasis by sedimentation. Tr. Roy. Soc. Trop. Med. and Hyg. 25:181–85.

Turner, J. H. 1951. Counting *Nematodirus spathiger* eggs in sheep dung. Proc. Helminth. Soc. Wash. 18:132–35.

Underhill, B. M. 1928. A simple method for concentration of parasitic eggs from feces. Univ. Pa. Vet. Ext. Quarterly, No. 32:4–6. *Also*: Jour. Am. Vet. Med. Assoc. 73 (n.s.26) :318–20.

United States Public Health Service. 1939. Supplementary basic technique for the recovery of protozoon cysts and helminth eggs in feces. (Preliminary communication.) U. S. Pub. Health Serv., Reprint 2014.

Vajda, T. 1922. A new method for detecting the eggs of parasites in feces. Jour. Am. Vet. Med. Assoc. 61 (n.s.14) :534–36.

———. 1927. Mételypeték kimutása bélsárbol dusito eljarással. Allatorv. Közlöny, Bd. 24, Nr. 1–3, S. 23–25.

———. 1940. Demonstration of liver fluke eggs by a new enrichment technique. (Trans. title.) Allatorv. Lapok. 63:147–49.

van den Berghe, L. 1936. Sur le polymorphisme des oeufs de *Schistosoma haematobium* et la présence d'oeufs du type bovin dans les infections de l'homme au Katanga (Congo Belge) . Bul. Soc. Path. Exot. 29:41–46.

von Ledden-Hulsebosch, M. L. Q. 1899. Makro- und microscopische Diagnostik der menschlichen Exkremente. J. Springer, Berlin.

Waddell, J. A. 1910. Hookworm in Virginia; its prevalence in certain Piedmont sections. Virginia Med. Semi-month. 14:546–49.

Waller, E. F. 1939. Pathology of *Eimeria arloingi* infection with observations on other coccidia of the intestine of sheep. Thesis, Iowa State College. 61 pp.

Ward, H. B. 1912. Means for the accurate determination of human internal parasites. Illinois Med. Jour., Oct.:1–18.

War Department. 1913. Methods of demonstrating and preserving ova of helminths. Office of Surgeon General, Wash., D. C. Bul. 1:9–37.

Wardle, R. A. 1932. On the technique of cestode study. Parasitol. 24:241–52.

Watson, J. M. 1947. A modification of the zinc sulfate centrifugal flotation technique for the concentration of helminth ova and protozoan cysts in faeces. Ann. Trop. Med. and Parasit. 41:43.

Weller, T. H. 1947. Diagnosis of *Schistosoma mansoni* infections: note on use of rectal scraper. Am. Jour. Trop. Med. 27:41.

———, and G. J. Dammin. 1945. An improved method of examination of feces for the diagnosis of intestinal schistosomiasis. Am. Jour. Clin. Path. 15:496–500.

———, ———, *et al.* 1945. The acid-ether centrifugation and the zinc sulfate flotation techniques as methods for the recovery of the eggs of *Schistosoma mansoni*. Am. Jour. Trop. Med. 25:367–74.

Wenrich, D. H. 1941. The morphology of some protozoan parasites in relation to microtechnique. Jour. Parasit. 27:1–28.

Wenyon, C. M. 1926. Protozoology: a manual for medical men, veterinarians, and zoologists. 2 vols. Baillière, Tindall and Cox. London, 1563 pp.

Westphalen, H. 1924. Milben in den Fäzes des Menschen. Deutsch. Med. Woch. 1:175.

Wharton, L. D. 1915. The development of the eggs of *Ascaris lumbricoides*. Phil. Jour. Sci. 10:19–23.

———. 1915. The eggs of *Ascaris lumbricoides*. Phil. Jour. Sci. 10:111–15.

White, M. J. 1914. Examination for hookworm ova. Pub. Health Rpts., Treas. Series. U. S. A. 29:8.

Whitlock, H. V. 1941. A new apparatus for counting small numbers of nematode eggs in faeces. Austral. Counc. Sci. and Indust. Res. Jour. 14:306–7.

———. 1943. A method of preventing the development of strongylid eggs in sheep faeces during transport and storage. Austral. Counc. Sci. and Indust. Res. Jour. 16:215–16.

———. 1948. Some modifications of the McMaster helminth egg-counting technique and apparatus. Austral. Counc. Sci. and Indust. Res. Jour. 21:177–80.

———. 1950. A technique for counting trematode eggs in sheep faeces. Jour. Helminth. 24:47–52.

Whitlock, J. H. 1938. Practical identification of endoparasites for veterinarians. Burgess Pub. Co., Minneapolis, Minn. 37 pp.

———. 1941. A practical dilution-egg-count procedure. Jour. Am. Vet. Med. Assoc. 98:466–69.

———. 1947. Illustrated laboratory outline of veterinary entomology and helminthology. Burgess Pub. Co., Minneapolis, Minn. 87 pp.

Whitney, L. F. 1936. A study of 1,000 fecal examination reports. Vet. Med. 31:104.

———, and G. D. Whitney. 1951. A study of canine fecal examinations. Vet. Med. 46:377–82; 418.

Widgor, M. 1919. A study of the fecal examinations of 1,000 imported dogs. Jour. Am. Vet. Med. Assoc. 56 (n.s.9) :189–91.

Widgor, M. 1920. Study of the character of the feces due to various foods in connection with anthelmintic investigation. Parke-Davis and Co., Res. Paper No. 171, 7:267.

Willis, H. H. 1921. A simple levitation method for the detection of hookworm ova. Med. Jour. Australia, 8:375–76. *Also* in 7th Ann. Rpt., The Rockefeller Foundation. Internat. Health Bd. for 1920.

Willmott, S., and F. R. N. Pester. 1952. Variations in faecal egg-counts in paramphistome infections as determined by a new technique. Jour. Helminth. 26:147–56.

Wilson, I. D. 1934. Sodium chloride vs. cane sugar for parasite egg flotation. Cornell Vet. 24:79–80.

Wood, W. A. 1932. A note on the size of the eggs of some species of sheep worms. Rpt. Dir. Inst. An. Path., Univ. Cambridge (1931), 2:220–22.

Wycoff, D. E., and L. S. Ritchie. 1952. Efficiency of the formalin-ether concentration technic. Jour. Parasit. 38 (Sec.2) :15–16.

Yaoita, S. 1912. Ein neues Verfahren zur Auffindung spärlicher Parasiteneier in Faeces. Deutsch. Med. Wchsr. 38 (33) :1540–41.

Young, B. P. 1929. A quantitative study of poultry coccidiosis. Jour. Parasit. 15:241–50.

Young, V. M. 1945. The staining of Protozoa in formalized stool specimens. Jour. Parasit. 31 (Suppl.) :7.

———, *et al.* 1951. A study of laboratory methods for diagnosing *Endamoeba histolytica* and their application to 5,048 persons from the Chicago area. Am. Jour. Digest. Dis. 18:126–30.

Zarrow, M., and H. Rufkin. 1946. Intestinal parasites diagnosed at an Army General Hospital in the South Pacific. Am. Jour. Med. Sci. 212:289.

Zawadowsky, M. M. 1929. The eggs of *Nematodirus spathiger* and the properties of their shell. (Trans. title.) Lab. Expt. Biol. Zoopark, Moscow, Trans. 5:251–54.

———, and S. N. Zvjaguintzev. 1933. The seasonal fluctuations in the number of eggs of *Nematodirus* sp. in feces. Jour. Parasit. 19:269–79.

Zeliff, C. C. 1947. (Laboratory diagnosis and laboratory techniques.) *In*: Manual of medical parasitology. Edwards Bros., Inc., Ann Arbor, Mich., pp. 109–48.

References for Section 2

EXAMINATION FOR MITES OF THE SKIN AND OF THE INTERNAL ORGANS

Armitage, F. D. 1936. A method for the preparation of mange mites for microscopical examination. Vet. Rec. 48:1404–6.

Ayres, S. 1930. Pityriasis folliculorum (Demodex). Arch. Dermat. and Syph. 21:19–24.

Baker, D. W. 1946. Barn itch. Parasit. Lab., Vet. Exp. Sta., Cornell Univ. 36 pp., 41 plates.

Baker, E. W., and G. W. Wharton. 1952. An introduction to acarology. Macmillan Co., New York. 465 pp.

Banks, N. 1915. The Acarina or mites. U. S. D. A., Report 108. 153 pp.

Bell, D. S., W. D. Pounder, B. H. Edgington, and O. G. Bentley. 1952. *Psorergates ovis,* a cause of itchiness in sheep. Jour. Am. Vet. Med. Assoc. 120:117–20.

Benbrook, E. A. 1929. Skin examination of domestic animals for evidence of parasitic mites. Iowa State Coll., Vet. Pract. Bul. 9:37–56.

Bingham, M. L. 1944. Some clinical diagnostic methods of use in conditions associated with animal parasites. Vet. Rec. 56:313–16.

Bishopp, F. C. 1942. Poultry mites. U. S. D. A., Yearbook:1055–61.

Buxton, P. A. 1921. External anatomy of the *Sarcoptes* of the horse. Parasit. 13:114–45.

———. 1921. On the *Sarcoptes* of man. Parasit. 13:146–51.

Cameron, A. E. 1924. *Sarcoptes* of cattle. Parasit. 16:255–65.

Carter, H. B. 1941. A skin disease of sheep due to an ectoparasitic mite, *Psorergates ovis* Womersly. Austral. Vet. Jour. 17:193–201.

Chandler, W. L., and D. S. Ruhe. 1940. *Pneumonyssus caninum* n. sp., a mite from the frontal sinus of the dog. Jour. Parasit. 26:59–67.

Coffin, D. L. 1945. (Examinations for ectoparasites.) *In:* Manual of veterinary clinical pathology. Comstock Pub. Co., Ithaca, N. Y., pp. 57–61.

Cooley, R. A. 1914. Killing small arthropods with the legs extended. Jour. Parasit. 1:105.

Cooper, K. W. 1946. The occurrence of the mite *Cheyletiella parasitivorax* (Mégnin) in North America, with notes on its synonymy and "parasitic" habit. Jour. Parasit. 32:480–82.

Cram, E. B. 1925. Demodectic mange of the goat in the United States. Jour. Am. Vet. Med. Assoc. 66 (n.s. 19) :475–80.

Crawley, H. 1922. A case of demodectic mange in a bull. Jour. Am. Vet. Med. Assoc. 61 (n.s.14) :441–43.

Davis, J. W. 1954. Studies of the sheep mite, *Psorergates ovis.* Am. Jour. Vet. Res. 15:255–57.

Ewing, H. E. 1929. A manual of external parasites. C. C Thomas Co., Springfield, Ill. 225 pp.

———. 1944. The trombiculid mites (chigger mites) and their relation to disease. Jour. Parasit. 30:339–65.

———, and A. Hartzell. 1918. The chigger mites affecting man and domestic animals. Jour. Econ. Entomol. 11:256–64.

Friedman, R. 1942. Biology of *Acarus scabiei.* Froben Press, New York, N. Y. 183 pp.

Hardenbergh, J. G., and C. H. Schlotthauer. 1925. Demodectic mange of the goat and its treatment. Jour. Am. Vet. Med. Assoc. 67 (n.s.20) :486–89.

Hearle, E. 1938. Insects and allied parasites injurious to livestock and poultry in Canada. Dominion of Canada, Dept. Agr., Publ. 604. 108 pp.
Hirst, S. 1922. Mites injurious to domestic animals. Brit. Mus. Econ. Series No. 13:1–107.

Imes, M. 1942. Mange in equines. U. S. D. A., Yearbook:476–81.
———. 1942. Mange of swine. U. S. D. A., Yearbook:734–40.

Koutz, F. R. 1953. *Demodex folliculorum* studies. II. Comparison of various diagnostic methods. The Speculum, 6:8–9; 23, 26, 55.
———, D. M. Chamberlain, and C. R. Cole. 1953. *Pneumonyssus caninum* in the nasal cavity and paranasal sinuses. Jour. Am. Vet. Med. Assoc. 122:106–9.

Leidy, J. 1872. On a mite in the ear of the ox. Proc. Acad. Nat. Sci. of Phila., p. 9.
Little, R. B. 1932. Demodectic (follicular) mange in cattle. Jour. Am. Vet. Med. Assoc. 80 (n.s.33) :922–26.

Martin, H. M., and M. J. Deubler. 1943. Acariasis of the upper respiratory tract of the dog. Univ. Pa. Vet. Ext. Quarterly, 89:21–27.
Miller, A. W. 1942. Sheep scab and its control. U. S. D. A., Yearbook:904–11.
Mönnig, H. O. 1938. Arthropod parasites. *In:* Veterinary helminthology and entomology. 2nd ed. William Wood and Co., Baltimore, Md., pp. 263–387.

Neveu-Lemaire, M. 1938. Recherche et étude des arthropodes. *In:* Traité d'entomologie médicale et vétérinaire. Vigot Frères, Paris, pp. 76–94.

Olsen, O. W., and F. K. Bracken. 1950. Occurrence of the ear mite, *Raillietia auris* (Leidy, 1872) of cattle in Colorado. Vet. Med. 45:320–21.
Olsen, S. J., and H. Roth. 1947. On the mite *Cheyletiella parasitovorax*, occurring on cats, as a facultative parasite of man. Jour. Parasit. 33:444–45.
Osborn, H. 1896. Insects affecting domestic animals. U. S. Dept. Agr., Div. Entomol., Bul. 5 (n.s.) . 302 pp.

Park, S. E. 1942. The cereal mite—a Pseudosarcoptes. No. Amer. Vet. 23:269–70.
Patton, W. S., and A. M. Evans. 1929. Insects, ticks, mites and venomous animals of medical and veterinary importance. Part I., Medical. H. R. Grubb, Ltd., Croydon, England. 786 pp.
Pillers, A. W. N. 1921. Notes on mange, and allied mites for veterinarians. Baillière, Tindall and Cox, London. 110 pp.
Price, E. W., and F. C. Bishopp. 1942. Mange of dogs. U. S. D. A., Yearbook:1174–79.

Reis, J., and P. Nobrega. 1936. Arthropodes parasitas. *In:* Tratado de doencas das aves. Inst. Biol., São Paulo, Brasil, pp. 353–85.

Sheather, A. L. 1915. An improved method for the detection of mange acari. Jour. Comp. Path. and Thera. 28:64–66.
———. 1943. The detection of intestinal protozoa and mange parasites by a floatation technique. Jour. Comp. Path. and Thera. 36:266–75.
Smart, J. 1948. A handbook for the identification of insects of medical importance. British Mus. (Nat. Hist.) , London. 295 pp.
Snyder, R. 1942. Cattle scab and its control. U. S. D. A., Yearbook:588–92.

Theobald, A. R. 1940. Parasitic skin diseases in dogs. Jour. Am. Vet. Med. Assoc. 97:139–44.

Warburton, C. 1920. Sarcoptic scabies in man and animals. Parasit. 12:265–300.
Wisseman, C. L., Jr., and S. E. Sulkin. 1947. Observations of the laboratory care, life cycle and hosts of the chicken mite, *Dermanyssus gallinae*. Am. Jour. Trop. Med. 27:463–67.

References for Section 3

THE DIAGNOSIS OF LICE INFESTATIONS

Babcock, O. G., and E. C. Cushing. 1942. Cattle lice. U. S. D. A., Yearbook: 631–35.

———, and ———. 1942. Hog lice. U. S. D. A., Yearbook:741–44.

———, and ———. 1942. Goat lice. U. S. D. A., Yearbook:917–22.

Baker, D. W. 1946. Barn itch. Parasit. Lab., Vet. Exp. Sta., Cornell Univ. 36 pp., 41 pl.

Bishopp, F. C. 1921. *Solenopotes capillatus,* a sucking louse of cattle not heretofore known in the U. S. Jour. Agr. Res. 21:797–802.

———. 1942. Some insect pests of horses and mules. U. S. D. A., Yearbook:492–500.

———. 1942. Poultry lice and their control. U. S. D. A., Yearbook:1048–54.

Chandler, W. L. 1917. Investigations of the value of nitrobenzol as a parasiticide with notes on its use in collecting external parasites. Jour. Parasit. 4:27–32.

Florence, L. 1921. The hog louse, *Haematopinus suis* Linné: its biology, anatomy and histology. Cornell Univ., Agr. Exp. Sta., Memoir 51, pp. 637–743.

Hearle, E. 1938. Insects and allied parasites injurious to livestock and poultry in Canada. Dominion of Canada, Dept. Agr., Publ. 604. 108 pp.

Herms, W. B. 1950. Medical entomology. The Macmillan Co., New York. 4th ed. 643 pp.

Kellogg, V. L., and G. F. Ferris. 1915. The Anoplura and Mallophaga of North American mammals. Leland Stanford, Jr., Univ. Pub's. (May 25.) 74 pp.

Lukens, W. R. 1921. To detect lice on horses. Jour. Am. Vet. Med. Assoc. 59 (n.s.12) :50.

Martin, H. M. 1932. Lice in dogs. Univ. Pa., Vet. Ext. Circ. 48:6–7.

Matthysse, J. G. 1946. Cattle lice, their biology and control. Cornell Univ., Agr. Exp. Sta., Bul. 832. 67 pp.

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